DOCUMENT RESUME

ED 309 065 SE 050 718

AUTHOR	Kelly, P. J., Ed.
TITLE	Field Work in Ecology for Secondary Schools in Tropical Countries. Science and Technology Education Document Series No. 30.
INSTITUTION	United Nations Educational, Scientific, and Cultural Organization, Paris (France). Div. of Science, Technical and Environmental Education.
REPORT NO	ED-88/WS/47
PUB DATE	88
NOTE	278p.
PUB TYPE	Guides - Classroom Use - Guides (For Teachers) (052)
EDRS PRICE	MF01 Plus Postage. PC Not Available from EDRS.
DESCRIPTORS	*Biological Sciences; Conservation (Environment); Developing Nations; Ecology; *Environmental Education; *Field Studies; Instructional Materials; Laboratory Equipment; Laboratory Procedures; Natural Resources; *Science Activities; *Science and Society; Science Experiments; Secondary Education; *Secondary School Science; Teaching Methods; Technology

ABSTRACT

One of the important aims of good biology teaching should be to encourage some basic understanding and appreciation of ecology. This understanding should include not only the scientific basis of ecology, but also its application for human welfare and, in particular, for rational exploitation and management of the natural environment and resources. Many of the problems faced by humanity are basically of an ecological nature: over-population; food production; deterioration of habitats; and pollution of air, water and soil. Thus, to make appropriate decisions related to aspects of human needs such as housing, agriculture, deforestation, industry, etc., an understanding of the scientific process of ecology is essential. It is to promote this understanding among biology teachers and their students that this document has been designed, with focus on simple experimental techniques in the field. The suggested projects or activities cover a variety of habitats, and individual species or their populations in tropical countries. Exercises include: (1) objectives; (2) curriculum links; (3) students' background; (4) students' preparation; (5) equipment; (6) methods; (7) teaching hints; and (8) follow-up. (Author/CW)

****
* Reproductions supplied by EDRS are the best that can be made *
* from the original document. *
****

"PERMISSION TO REPRODUCE THIS MATERIAL IN MICROFICHE ONLY HAS BEEN GRANTED BY

Susanne Adlung

TO THE EDUCATIONAL RESOURCES INFORMATION CENTER (ERIC)."

U.S. DEPARTMENT OF EDUCATION
Office of Educational Research and Improvement
EDUCATIONAL RESOURCES INFORMATION CENTER (ERIC)

This document has been reproduced as received from the person or organization originating it.

☐ Minor changes have been made to improve reproduction quality.

• Points of view or opinions stated in this document do not necessarily represent official OERI position or policy.

Field Work in Ecology for Secondary Schools in Tropical Countries

BEST COPY AVAILABLE

ED309065

SE 050 718

Cover photos

1. Photo Unesco/Paul Almasy
2. Photo UNATIONS
3. Photo Unesco/D. Bahrman
4. Photo rights reserved

Science and Technology Education

Document Series No. 30

Field Work in Ecology for Secondary Schools in Tropical Countries

Edited by Professor P.J. Kelly

Division of Science
Technical and Environmental
Education

ED-88/WS/47

UNESCO

Paris, 1988

P R E F A C E

The document series has been established as part of Unesco's Science and Technology Education Programme to encourage an international exchange of ideas and information in the field. The present volume was produced under the auspices of the Commission for Biological Education of the International Union of Biological Sciences (CBE-IUBS), under contract with Unesco.

One of the important aims of good modern biology teaching must be to encourage some basic understanding and appreciation of ecology. This understanding must include not only the scientific basis of ecology, but also its application for human welfare - in particular for rational exploitation and management of the natural environment and resources.

Many of the problems which we face today and may continue to face tomorrow are basically of an ecological nature: over-population, food production, deterioration of habitats and pollution of air, water and soil. Thus, to make appropriate decisions related to aspects of human needs such as housing, agriculture, deforestation, industry etc., an understanding of the scientific process of ecology is essential.

It is to promote this understanding among biology teachers and their students that the present document has been designed, with focus on simple experimental techniques in the field. The suggested projects or activities cover a variety of habitats, individual species or their populations in tropical countries.

Efforts have been made to model each exercise with the following sub-headings:

- objectives
- curriculum links
- background of students
- students' preparation
- equipment
- strategy and methodology
- teaching hints
- follow-up.

It is hoped that biology teachers will try various activities in different habitats and on the basis of their experience will send their comments and feedback to Unesco's Division of Science, Technical and Environmental Education.

The format of this document was developed in a workshop which was convened and organized by Dr. Twee Hormchong, Director, Bangsaen Institute of Marine Science, Chonburi, Thailand on behalf of the CBE-IUBS; 18-25 January 1987. The Editor is Professor P.J. Kelly, Professor of Education, University of Southampton, U.K.

The contributions and co-operation of the authors and the editor in the preparation of this book, the help from Mr. J.P. Pritchard in preparing the issustrations and the care and hard work of Mrs. Annette Bailley in the preparation of the manuscript are gratefully acknowledged.

The opinions expressed in the following pages are those of the authors, and not necessarily those of Unesco.

CONTENTS

		Page
Chapter 1	Introduction	1
Chapter 2	Ecological Principles	5
Chapter 3	Human Exploitation and Management	11
Chapter 4	Ecological Techniques	17
Chapter 5	Ethological Methods	61
Chapter 6	Practical Activities	69
	The Marine Environment	71
	Humid Forests	161
	Savanna	225
	Deserts	249
	Arid and Semi-Arid Lands	271

Bibliography

CHAPTER 1

INTRODUCTION

The term tropics refers to that part of the earth's surface between latitudes 23.5°N (Tropic of Cancer) and 23.5°S (Tropic of Capricorn). Its limits are set by the turning points in the seasonal movement of the earth relative to the sun. On June 22nd at noon the sun is vertically above the Tropic of Cancer. On December 22nd at noon it is above the Tropic of Capricorn.

Within this area there is a wide variety of environments. Mainly they are hot although in mountainous areas the effect of altitude results in the icy conditions of higher latitudes. On land the natural flora and fauna ranges from those of the dense, humid forests containing, it is estimated, three-fifths to three quarters of the world's species, to the barren, virtually lifeless deserts. In relation to this, rainfall varies from over 5000 mm per annum to nothing. The warm waters of the tropical sea-shore and ocean provide the circumstances for the richest flora and fauna of any aquatic habitat.

Compared with the temperate region the tropics cover over 20 million Km^2 more area and, besides containing many more animal and plant species, is populated by three times as many human beings (soon to be two-thirds of the world population). Many of its countries are poor and have considerable shortages of food and energy and problems arising from excessive exploitation of natural resources. It is an area of immense natural riches and yet, in a scientific sense, is relatively unknown and economically among the least productive of all inhabited regions on the earth.

It is now an accepted fact that the solution to many of the difficulties that countries in the Tropics face are to do with their ecology - the relationships between the living world, including man, and the environment. We have witnessed dramatic examples of the perils one faces when our understanding of ecology is inadequate or ignored: the thousands of square kilometres of once productive savannah turned into desert through over-grazing, the destruction of forests by the excessive use of slash and burn methods of shifting agriculture, over-fishing in rivers and the sea, and the haphazard use of insecticides to the point when plant production becomes uneconomically dependent on them. Such catastrophes can be avoided if we have sufficient knowledge of ecology and apply it wisely.

The study of ecology can be considered to have two major perspectives, scientific and utilitarian. In making this distinction the role of science is conceived as the pursuit of knowledge for its own sake whilst the term utilitarian refers to the application of that knowledge for human welfare and, particularly in relation to ecology, the way we exploit and manage our environment. They are activities which, in reality, are linked closely, but the distinction is important because it reminds us that from science we obtain the basic ideas which we can adapt in a variety of ways to deal with practical issues.

Particularly it reminds us that a sound scientific understanding of ecology is essential to the success of a range of major human endeavours such as agriculture, forestry, fishing, conservation and pollution control.

Education has a major responsibility in this respect. It is through education that we produce the scientists who increase our knowledge of ecology and educate those who put it to practical use. At the same time through education we can enable citizens, and particularly their leaders who make the decisions in public life, appreciate the importance of ecology to human welfare.

In recent years ecology has become a more prominent topic in the school curriculum throughout the world. Usually it features as part of a biology course although it is also to be found in integrated science, geography and social studies courses and in broader programmes of environmental education. However, its impact is often diminished because the teaching of ecology lacks reality. Too often this consists of mere descriptions of organisms in their environment without providing students with direct experience of studying ecology in action.

In tropical countries this state of affairs is understandable. Most of our scientific understanding of ecology comes from temperate regions, most of the guides to practical ecology are written for temperate habitats. There is a need for publications which can provide teachers, especially biology teachers, in tropical countries with ideas for easily managed practical work in the field. This book aims to help in meeting this need.

It is not a textbook on ecology. For a detailed theoretical background to the practical activities described here, and which form the greater part of this book, you should consult some of the publications listed in the bibliography.

However, in Chapters 2 and 3 are provided elementary reviews of the fundamental principles of ecology and of the major aspects of man's exploitation and management of the environment which have ecological relevance. Such frameworks of ideas can be useful in providing a holistic view of a topic and will remind students of essential concepts without their getting lost in detail. These need to be in their minds continually because the secret of successful field studies in ecology is to ensure that they relate to basic ecological understanding.

In Chapter 4 a number of practical techniques which can be used in field work in a wide variety of habitats are outlined. They are referred to in a number of investigations later and will be useful when you wish to design your own activities.

Chapter 5 is concerned with methods used for the study of animal behaviour (ethology). Whilst animal behaviour is frequently taught separately from ecology there are many connections between the subjects and here we take the view that a number of aspects of animal behaviour should be treated as part of ecology.

Chapter 6 is the major focus of this book. In it is a collection of practical fieldwork investigations provided as examples of the type we might aim for in secondary schools. Each investigation is written so that teachers can easily adapt it for their own use. From the collection as a whole teachers should be able to pick up sufficient ideas to plan a programme of field investigations to suit their own circumstances.

The collection will have served its purpose if it encourages and enables teachers to take up field work, and stimulates teachers and ecologists to combine to produce better and more relevant schemes for their own countries.

CHAPTER 2

ECOLOGICAL PRINCIPLES

The main ideas or principles of ecology can be easily understood by considering any place (called a habitat) where animals and plants are to be found. In this review we will examine them under four headings.

A. Diversity and distribution
B. Relationships
C. Change and equilibrium
D. Movement and behaviour

DIVERSITY AND DISTRIBUTION

One of the first things that will attract your attention in the habitat is the diversity and distribution of the organisms. There will be different kinds of plants and animals and they will be distributed in different ways within the habitat.

To study them we will first have to identify the organisms and to do this we must know something about the ways they are classified. Taxonomic keys will help us do this. In these organisms are placed in groups according to their similarities and differences. The groups are called phyla, classes, orders, families, genera and species. Phyla contain many different organisms but they all have a few features in common. Species are groups containing the least number of different organisms and they have many features in common. The other categories contain decreasing differences in the order listed above from phyla to species. Where we place an organism is decided by biologists who specialise in the study of classification (taxonomists) and we have to consult their books to determine the position of an organism.

Incidentally, the scientific name of an organism is provided by its genus and species. For example, Homo sapiens is that of human beings. Homo refers to our genus, sapiens refers to our species.

In ecological study it is sometimes valuable to construct other forms of classification. For example, we may wish to group animals according to the food they eat and so we could classify them as carnivores (meat eaters), herbivores (plant eaters) and omnivores (those that eat plants and animals).

Within the habitat organisms will be distributed both horizontally - as with most plants growing in the ground - and also vertically - as, for example, with those plants which live at different levels on trees and; of course, most species of birds and insects are spread out in the air, not just on the ground.

Having observed the distribution we will, no doubt, ask questions about the reasons for it. Is it to do with aspects of the environment? To answer this we will need to study the distribution of physical factors such as light, temperature and chemicals. This will in turn lead us to think about how the organisms survive in the habitat and hence the forms of adaptation to their environment which enable them to do so.

We will ask questions also about how the organisms got to the habitat (colonization) and how they are subsequently dispersed throughout it.

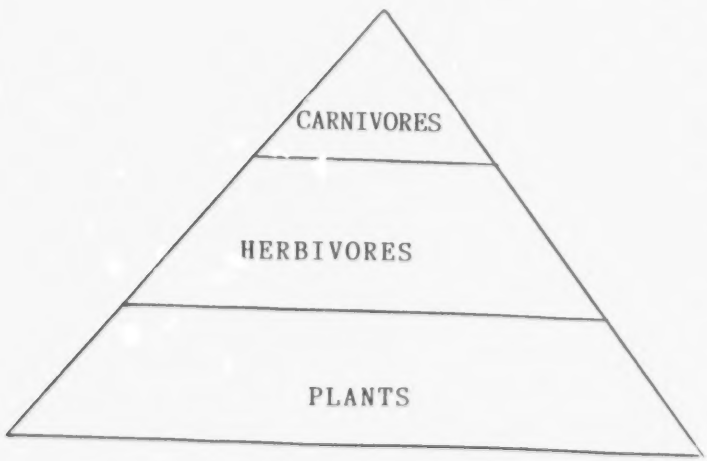
From all this a picture of a complex set of relationships - between organisms, and between organisms and their environment - will emerge.

RELATIONSHIPS

One of the simplest relationships is the food chain. Plants (producers) in the habitat will be eaten by animals (herbivorous consumers) and they in turn by other animals (also called consumers but, of course, carnivorous). In this way their lives are connected like a chain. This relationship effects the number of organisms. There will be a larger number of plants (or a smaller number of large plants) compared with the herbivores and, in turn, these will tend to exceed the carnivores in number. This relationship is depicted by a pyramid of numbers.

FIGURE 1

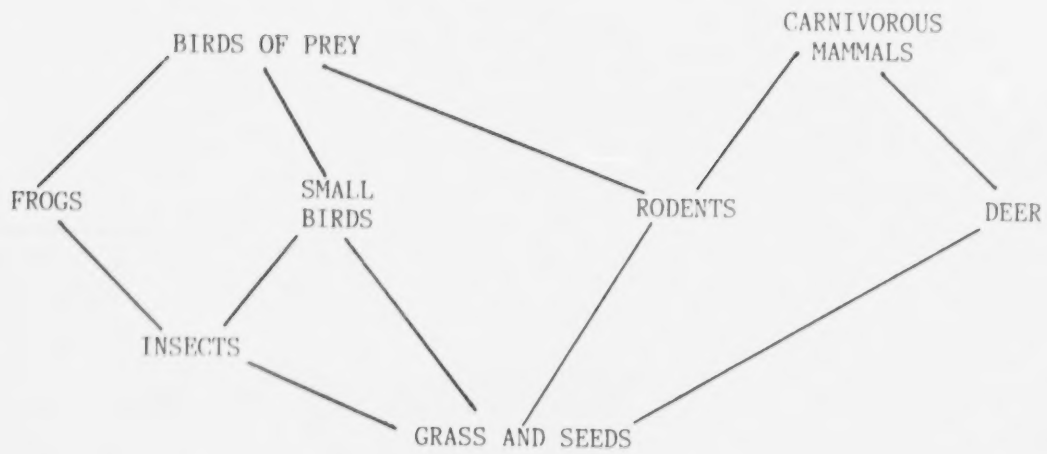
PYRAMID OF NUMBERS



However, close observation and analysis will reveal that things are somewhat more complex than this. Different food chains are linked to form food webs as when more than one type of herbivore feed on the same plant and they are fed on by different carnivores.

FIGURE 2

AN EXAMPLE OF A FOOD WEB



Through food webs the substance of organisms (biomass) and energy flows from one location to another.

There may also be examples of other relationships between organisms such as those of parasitism (one organism living and depending on another) and symbiosis when two organisms live closely together to the advantage of each other.

Organisms also have close relationships through their interaction with the physical environment. Plants, for instance, replenish the air with oxygen through photosynthesis which is used by animals in respiration. From respiration comes carbon dioxide which, of course, plants can use for photosynthesis. There are many examples of such cycles in a habitat. They are the means by which chemicals move from one location to another.

All these relationships put together in a habitat make up an ecosystem. Understanding ecosystems is the main focus of ecological study.

CHANGE AND EQUILIBRIUM

An ecosystem does not remain static. A piece of waste land colonised by a few plants and animals will be found to change over time. Communities of small organisms will be followed by more complex communities containing usually some larger plants and animals. Eventually, by a process of succession an ecosystem will be established which is more balanced and thus in equilibrium. This will contain the climax community which will tend to remain the same unless major changes in the environment upset it. Then the process of succession will commence again but, possibly, in a different form.

Even within a climax community there will be some changes, although they tend to balance out. During their life cycles some organisms change considerably; caterpillars change to butterflies, for example. This produces different relationships as when the different stages require different food.

Within the ecosystem organisms compete against each other for resources; some survive and others do not. If populations of herbivores get too large the number of plants is reduced. This in turn will decrease the number of herbivores until the plants recover. In such cases cycles of population size occur over time.

Changes also occur in ecosystems between day and night. They have a diurnal cycle. Most animals feed during the day and rest at night. However some, such as bats and many moths, feed at night and rest during the day. Over each period of 24 hours one set of relationships between organisms will succeed another as night and day alternate.

Over greater periods of time an ecosystem is changed by evolution by which, through the process of natural selection, new varieties of organisms are produced.

MOVEMENT AND BEHAVIOUR

In observing a habitat it is important, at times, to stay still, for it is then that we can appreciate the amount of movement that is occurring. Change in ecosystems is frequently associated with movement. Plants get dispersed through, for example, being carried by wind or insects. The wind, also, can carry top-soil away and prevent plants growing. Fixed creatures like marine barnacles are dispersed when their free-floating larvae are carried away by water currents. In these and many other ways movement of organisms and the physical environment occurs both within and between ecosystems.

The behaviour of animals (an important aspect of ecological study), involves movements often of great significance to ecosystems. The mass migration of antelopes in the savannah and fish in the sea are examples of this. Yet the less obvious short-range movements of animals for feeding, mating, protection and other needs can also effect the balance of an ecosystem.

And we should not forget that plants can move. Mostly they do so through growing and the effects on the ecosystem are difficult to detect but, for example, in humid forests climbing plants are prominent and plentiful members of communities providing a habitat for a diverse variety of other organisms.

This brief account provides a framework of ideas on which the study of ecology can be based. It is not complete but it is sufficient to enable students at the secondary level of education to interpret their field work in terms of fundamental ecological principles.

CHAPTER 3

HUMAN EXPLOITATION AND MANAGEMENT

Earlier in Chapter 1 two main reasons for studying ecology were suggested. One concerned its scientific value. The other - and, today, probably the most important reason - was the value of ecological knowledge in enabling us to look after our physical and biological environment sensibly. This involves us in considering the advantages and disadvantages of the ways we exploit our environment and how we can manage it to be of long-term benefit. Exploitation and management are, thus, essential topics in a course in ecology and consequently we should endeavour to organise field work studies that relate to them.

In this review aspects of these topics are dealt with under four headings.

A. Exploitation of natural habitats
B. Production
C. Pollution
D. Conservation

EXPLOITATION OF NATURAL HABITATS

When a forest is cut down it can take many years to be replaced. In fact it may never return if, as has happened in some parts of the world, the wind, rain, or both, sweep the top soil away and a desert remains. Yet, you may ask, is it not reasonable that people should use the wood for burning (to keep themselves warm and cook their food) or for building their homes?

This is the type of problem we have to face when we exploit natural habitats. How can we ensure that our use of them, which to some extent is a necessity, will not eventually result in our losing them?

Exploitation of habitats occurs in a number of ways. Organisms can be removed from the habitat as, for example, with fishing or tree-felling. Organisms can be damaged by, for example, the removal of parts as when branches are broken off for firewood or grass cut for fodder. The introduction of new organisms into habitats is also a form of exploitation, and can have harmful effects. The rabbit introduced into Australia in the last century quickly became a pest by eating enormous amounts of vegetation and so upset the ecological balance. Diseases in farmstock can arise from the careless introduction of infected animals and plants.

Exploiting the physical environment can have effects too. The removal of soil and minerals will interfere with the chemical cycles on which organisms depend. Altering the flow of a river by building a dam can cause both flooding in its upper regions and drought in its lower parts. Either way plants and animals will be affected.

Such occurrences result from exploitation when ecological principles are not taken into account. There are, fortunately, many examples of sound ecological exploitation such as with fishing when the amount of stock that can be taken is limited by regulation, with forestry when the felling of trees is balanced by a carefully worked out system of replacement, and with national parks where the needs of tourists are managed so as not to upset the ecosystem they have come to enjoy.

PRODUCTION

The most obvious way in which people expolit nature is by the replacement of natural ecosystems by man-made ones. Cattle ranches and rice fields are such man-made ecosystems; so is a plantation or a fish farm, for instance. Also, the cities and villages in which we live can be considered as ecosystems. And in all such cases the same fundamental ecological principles apply as they do to natural systems.

A major reason why man-made ecosystems come about is the need for production - of food, of energy, of materials for clothing and shelter, and so on. Because of this man-made production ecosystems tend to differ from natural ones in a number of ways.

First, the balance of a production ecosystem is, to a large extent, controlled through the use of artificial methods which depend for their effectiveness on decisions made by people. For instance, on farms the addition of fertilisers is often necessary to maintain chemical cycles resulting in high yields of crops. However, the excess use of fertilisers can be harmful both to the crops and organisms living with them which are necessary for their survival. How well the farm ecosystem functions depends essentially in this respect, on decisions made by the farmer about the amount of fertiliser that is used.

This example of the use of fertilisers also illustrates another difference between production and natural ecosystems. For production purposes parts of the ecosystem are continually being removed and have to be replaced. Farm crops are regularly harvested and have to be replaced by planting or seeding. The chemicals they have taken from the soil have also to be replaced.

A third difference is that a production ecosystem is influenced considerably by economic and social factors. The extent of production and the methods employed will depend on the demand for the products and the costs involved. Besides an ecological balance there has also to be an economic balance. People's values and beliefs will also be influential as when religious ideas effect the way people work and how they treat animals and plants.

Scientific knowledge also can effect production systems. Genetics helps in producing the most productive organisms, physiology and allied subjects help to determine their environmental requirements and, above all, ecological knowledge helps to ensure that there is an appropriate balance within the food webs, chemical and energy cycles, and other aspects of the ecosystem.

POLLUTION

Pollution occurs when something is added to an ecosystem which has a harmful effect. Among well-known examples are marine oil pollution, which arises when oil released from ships and other sources forms a layer over water that prevents the exchange of gases between the water and the air on which the ecosystem depends and kills sea birds by clogging their feathers and preventing them flying. 'Acid rain' is another example. This results from the release of sulphur dioxide and other gases into the air by factories which is then deposited in rain as sulphuric acid and similarly harmful substances causing damage to forests and aquatic life. Nitrate pollution occurs when nitrates used for fertilising soil are carried away when there is heavy rain to rivers and lakes where they accumulate in large quantities and become poisonous or result in such excessive growth of algae and other plants that the balance of the aquatic ecosystem is seriously upset.

Most forms of pollution arise from human activities but some arise naturally. In parts of the world, for example, volcanic activity, from time to time, releases poisonous chemicals from deeper layers of rocks which destroy animals and plants and can make lakes sterile. Also, for example, natural changes in geological formations sometimes slow down rivers or block them completely. Decaying plant material then accumulates making the water stagnant. The form of the ecosystem will certainly be changed and, sometimes, plant and animal communities are destroyed.

An understanding of man-made and natural pollution and their effects is necessary for the successful management of ecosystems. We need also to understand that the effect of a pollutant may be indirect. For example, insecticides are valuable in controlling pests that destroy crops. However, because other animals that feed on the insects may also be harmed, an insecticide may have secondary i.e. indirect effects which upset the ecological balance. This is the case with the insecticide DDT. It is passed from insects to the birds that eat them and then to birds of prey which feed on them. The DDT chemical causes the birds of prey to lay thin-shelled eggs and consequently their offspring do not survive. This decreases their population which, in turn, increases the number of herbivorous animals on which they feed. Consequently, and paradoxically, extra damage is caused to crops.

As with so much to do with ecosystems the effects of pollutants are not necessarily simple. Pollution of ecosystems has to be appreciated as a complex phenomenon but if you understand basic ecological principles it is surprising how easy it becomes to unravel the complexities.

The important thing about pollution is that we should know when it occurs and how great it is. For this we need to have ways of detecting and monitoring pollutants in ecosystems. It involves using tests over periods of time for chemical and physical aspects of the environment and changes among organisms.

Sometimes we can use features of organisms as indicators of pollution. The flat forms of lichens are examples. They survive in polluted air when the bushy forms are unable to.

When pollution has been detected the need then is to eradicate it and restore the balance of the ecosystem. This can be done by finding the source of the pollutant and stopping it; something which often requires legislation as well as scientific evidence. Sometimes it is necessary to find a substitute for a pollutant which, whilst it has disadvantages, has advantages which need to be retained. The introduction of animals which feed on insect pests instead of using DDT is an example, but, of course, you have to be careful that the substituted method does not cause problems as well. It is not always possible to foresee the effects of manipulating an ecosystem. We have to be careful and, above all, be continually on the alert to detect unsuspected outcomes.

CONSERVATION

Conservation is the term we use for the maintenance of special areas in as natural a condition as possible. There is now a world-wide effort to set aside habitats of all different kinds in order to maintain the environment and organisms they contain. There are four main reasons for this. The first is that of survival. The loss of natural ecosystems is a loss of natural resources on which we ultimately depend. The loss could also be catastrophic. At the current level of loss of plants, especially those of the tropical forests, a 'greenhouse' effect is being produced through the accumulation of the carbon dioxide in the atmosphere which is no longer taken in by the plants for photosynthesis. It is estimated that this could eventually increase the earth's surface temperature sufficient to melt part of the polar ice cap and flood coastal towns throughout the world. Through conserving habitats such problems can be avoided.

The second reason is scientific. Through maintaining a diversity of species in conservation areas we are also maintaining their genes. This provides scientists with the raw materials by which, through genetical research, new and beneficial varieties of plants and animals of value in agriculture, medicine and other ways can be developed. Indeed, conservation areas provide scientists with natural laboratories for many fields of research including, of course, ecology - about which we still have much to learn.

Conservation areas can also serve as outside classrooms, places where students and the public can learn at first hand about the natural world and ecology in particular. Hopefully this educational function will prosper as a result of the book you are now reading!

The fourth reason is that of enjoyment. Natural habitats provide people with the pleasure of their beauty and interest and, at times, a welcome change from the materialistic pressures of urban life. In most cultures of the world one finds, at least to some degree, a valued respect for human relationships with the natural world. Very often it is this that provides the motive for the establishment of conservation areas.

For whatever reasons conservation areas are maintained they require careful management. This needs to be based on sound ecological principles and in this respect it is important to recognise that conservation is not exactly the same as preservation although that is an aspect of it.

Sound conservation management recognises that change continually occurs in nature and that it is necessary to maintain the capacity for change. To conserve a habitat will, of course, preserve the species living there to some extent. However by normal ecological and evolutionary processes they will inevitably change over time. Even successive generations of the same species will show at least small differences. Conservation is concerned with preserving ecosystems.

CHAPTER 4

ECOLOGICAL TECHNIQUES

Ecological investigations require both intellectual and practical techniques. The former are essentially those of scientific problem-solving: devising questions that can be answered by scientific means, making careful and objective observations, keeping accurate records, devising testable hypotheses for experiments, utilising appropriate experimental designs, analysising data carefully - using quantitative methods where appropriate, and drawing conclusions in a logical manner.

A number of the practical techniques used in ecology will also be encountered in other aspects of science but of specific relevant to ecology are techniques of collection and measurement as applied to organisms and the physical environment. In the practical investigations that follow in Chapter 7 details of specific uses of some of these techniques are given. Here techniques of general application are outlined and in Chapter 5 most of the apparatus required for them are illustrated.

COLLECTING APPARATUS

The following pieces of apparatus will have a wide use in a school ecology course.

HAND LENS - of medium power (x8 or x10), used for recognition and identification of small organisms and small structures in larger organisms.

BULB-PIPETTE - both wide and narrow mouthed; for sucking up and transferring small organisms to storage containers. (Figure 5, page 49)

FORCEPS - for handling medium sized, hard bodied organisms.

PAINT-BRUSH - for handling delicate soft-bodied organisms e.g. flat worms and aphids.

WIDGER - an all-purpose tool for digging out plants, chipping off organisms from rocks, levering open crevices, excavate rotting wood etc. (Figure 4, page 49)

POOTER (aspirator) - for collecting and transferring insects, arachnids and similar sized organisms. This can be constructed from a specimen tube (3" x 1"). (Figure 3, page 49) The mouthpiece is placed at the end of a flexible tube of suitable length so that insects can be captured at a comfortable viewing distance. The free end of the suction tube is covered with a piece of muslin to protect the operator from swallowing the collected specimens.

SPECIMEN TUBES - for storing specimens. There are assorted sizes including some of the same size as the pooter tube so that it is easy to replace the full tube with any empty one. A particularly useful size is 2 in. x 1 in.

SIEVE - an ordinary all-metal kitchen sieve is useful for sifting soil, mud, sand or pond water for small organisms.

SCREW-TOPPED JARS - for collecting and transporting organisms, particularly aquatic ones.

POLYTHENE BAGS - different sizes for carrying various items including soil, weeds and aquatic animals.

ENAMEL DISH - for sorting aquatic or terrestrial organisms.

KILLING JAR - This can easily be made from a used wide-mouth jam jar or from an empty cigarette tin depending on the size of organisms being dealt with. Place a layer of about 1 - 2 cms. of a mixture of potassium cyanide and plaster of paris at the bottom of the container and cover it with a filter paper. Make sure that the cover is tight fitting. If cork is used as such, it should be dipped in molten paraffin so as to block the pores. The gas issuing from the mixture is poisonous and should not be inhaled. Alternatively chloroform soaked in cotton wool might be used. The use of a killing jar must be controlled by the teacher bearing in mind moral sensitivities that may be involved in relation to killing animals. (Figure 8, page 50)

NETS - These may be divided into three main types: kite net, sweep net and water net; with much variation in shape, size and material. Those for use in water tend to be smaller-mouthed and are made of stronger materials. The principles of collecting with nets, however, are the same as for air. All nets should be dried and cleaned after use, mending wear-and-tear as soon as possible.

COLLECTING TECHNIQUES

(a) Flying Insects

For catching diurnal insects in flight a short-handled (60cm) kite net is useful. It can be made from a light flexible frame of cane or bamboo and a suitable piece of mosquito-netting or muslin cloth, preferably of dark green colour (See Figure 9, page 50). The length of the bag should be about twice as long as the diameter of its mouth to stop the larger and more powerful insects

from escaping. To catch an insect, the net should be used with a rapid sweeping movement ending with a twist of the wrist to close the mouth of the net.

(b) Organisms in Herbage and Trees

Grass-hoppers, beetles, larvae, spiders etc. can be collected by sweeping a fairly robust sweep net, mouth first, through grass or soft herbage, so that the dislodged organisms fall into the bag. The depth of such a bag need not be as much as with the kite net. The mouth of the bag can be round or triangular but its bottom should be round. The handle should not be too long and unwieldy (Figure 10, page 51)

A beating tray can also be used for the same purpose. It comprises a piece of strong cloth, preferably black or dark green in colour, stretched on a light collapsable frame. (An umbrella can be used as a beating tray.) The tray is held under bushes or tree branches which are then beaten by a stick, allowing the dislodged organisms to fall on to the tray. The fallen organisms can be collected with an aspirator (pooter).

(c) Water Organisms

Some aquatic species such as Molluscs are comparatively sedentary and are found clinging to the surface of vegetation or stones and hence are best collected by hand using a scalpel or pen-knife. Some of the mobile species, however, require the use of some kind of net. A large variety of aquatic nets is now available.

The smaller of these are operated by hand, while the larger ones can be towed behind a boat. Since fieldwork at school level can involve collecting for a variety of purposes three main types of nets are described below:

(i) Plankton net: This is a small net with a circular mouth of cane or a stout wire frame, about 15 cm in diameter. The bag, made of closely woven nylon material or bolting silk, black in colour, tapers to a point through which a rimmed glass tube or a small bottle is secured for the collection of plankton. The size of the organism retained by the net is the function of its mesh. Thus 60 and 180 meshes per inch are recommended for the collection of zoo-plankton and phytoplankton respectively. A plankton net will collect efficiently if moved at a low speed of 1 - 2 miles per hour. For collection in shallow ponds, small plankton nets can be fitted to ordinary hand-net frames. (Figure 11, page 51)

Larger swimming or free-moving organisms can be collected by a tow-net which is similar in overall dimensions to the plankton net but with a larger mesh. Unlike the plankton net, it is moved with greater speed.

(ii) **Drag-net:** A drag-net or dredge is a framed bag with a handle. The material of the bag should be such as to let through fine sand but not animals visible to the naked eye. It can be used with moderately fast to-and-fro sweeps.

A drag net can also be designed for hauling over a surface. For use on a grassy or rough surface the frame should have runners like a sledge so that the net can be drawn along with the mouth of the bag facing forwards. (Figure 12 , page 52) This type of net is useful for quantitative sampling of caterpillars or other insect larvae which are readily dislodged from vegetation.

A drag net can also be used as a static net in a stream to collect organisms deliberately dislodged from a convenient position upstream.

(iii) **Trawl net:** This is a heavy duty drag net for use under water. It has a large conicalnet bag attached to a wide circular or triangular frame (Figure 13, page 52) It can be trawled by a rope from a boat or by two people on opposite banks of a stream. It is used for catching larger aquatic animals.

(d) **Nocturnal Animals**

Many nocturnal animals are best collected by traps using ultra-violet light or a mercury-vapour (M.V.) lamp. Some examples of these are -

(i) **Simple Light Trap:** This consists of an illustrated white sheet as shown in Figure 14, page 53 . Being attracted to the light, insects fall on the sheet from where they can be picked up for study.

(ii) **Moth Trap:** One of the more efficient forms of the moth trap can be made from a wooden box about 60 cm square and 20 cm deep with a bottom of perforated zinc to allow drainage of rain water if any. The bottom is strewn with cardboard fragments or similar material to receive the trapped and fallen insects. The top is covered with two overlapping pieces of glass sheet with a vertical opening of about 2.5 cm. One of the glass sheets should be so fixed that it can be slid in or out when needed. The metal stem carrying the M.V. lamp should project upwards above the glass-sheets (Figure 5, page 53). Moths, when attracted by the trap, will slide down to settle on the cardboard pieces from where they can be collected.

(iii) **An Improvised Light Trap:** This consists of a hand torch enclosed in a water-tight jar and placed inside a plastic drainpipe (Figure 16, page 53). It is covered with a wire-gauze funnel at one end (in-let) and a detachable wire-gauze lid (out-let) at the other. This design can also be used without light to trap small fish or other organisms from a stream.

(iv) **Pitfall Trap:** These are good for collecting crawling or running insects. A jam-jar sunk into the ground can serve the purpose. The mouth of the jar should be level with the soil and protected by a roof of stone or wood. The ground should slope away from the mouth of the jar which should contain bait to attract the insects. Such traps should be visited frequently and any trapped insect should be collected and stored. (Figure 17, page 53)

(e) **Extraction of Organisms from Soil, Leaf-Litter and Moss**

Many soil organisms are microscopic in size and the special methods for their collecti n are beyond the scope of this book. However, a great variety of larger invertebrates live in wood and litter, piles of loose moss and moss growing on the ground, accumulated debris brought by rivers in floods or left high up on the shore by the receeding tide. Such organisms can be collected by various methods. While none is a hundred per cent efficient, there are some which are sufficiently reliable and can be used for comparative estimates of populations in different habitats. Thus by studying samples using 'handfuls of litter' or 'cans of soil' as units an approximate idea of the density of individuals in the community can be made.

An opaque white water-proof sheet is useful for examining soil and leaf-litter in the field. Some of the more specialised methods include the use of funnels, chemicals and flotation and are described below:

(i) **Funnel Methods**

The Berlise-type funnel depends on the simple principle that various soil and litter organisms tend to move away from heat. Loose soil and litter is placed in a sieve with four holes to the square inch. The sieve is fitted in a large metal funnel surrounded by a water jacket heated from below. A jar containing suitable liquid (70% alcohol or 4% formalin) is placed under the funnel for direct preservation. The warm air inside the funnel rises and circulates through the material in the sieve and drives the organisms downward into the collecting jar. (Figure 18, page 54)

Modern modifications of the Berlise method are mostly of the Tullgren pattern employing both heat and light for extraction (Figure 19, page 54). To be efficient the funnel must be smooth and must not produce condensation which can trap more delicate organisms and thus stop them from reaching the bottom. The apparatus can easily be improvised by using an elongated steep cone made from smooth, thick paper or cardboard instead of metal. The soil or litter sample can be placed in a suitable sieve or wire-gauge fitting nicely in the funnel. It can then be heated from above by a 40 - 60 watt electric light bulb suspended from a lid over the funnel.

Organisms, when activated, move downwards to fall into the collecting receptacle. If the organisms are required alive for study, blotting paper wet with water should be used in the receptacle instead of a liquid preservative. Predators such as centipedes should be collected as soon as they fall out otherwise other organisms will be consumed.

The period of heating required varies with the amount and nature of the soil. Light dry soils may need about 24 hours, while heavier, wet ones require 2 - 3 days.

Since nematodes are heavier than water, the Baermann funnel is used for their extraction (Figure 20, page 54). With this the soil sample is placed in a muslin bag and submerged in water. The nematodes leave the soil and sink to the bottom of the funnel from which they are collected after twelve hours by drawing off the bottom half-inch of water. Gentle overhead heating can increase the extraction efficiency.

(ii) **Chemical Extraction**

This method is used, for example, for sampling earthworm populations. A weak solution of potassium permanganate or an aqueous solution of formalin (25 ml of 40 per cent formalin per gallon of water) is sprayed with a watering can on the area being studied. Earthworms come to the surface in about half an hour. This method is used as an alternative to digging up the soil but is not quite so efficient. A few individuals often do not respond to the effects of the chemical.

(iii) **Flotation Method**

This is specially used for collecting small arthropods, insects and larvae which float readily in liquids with specific gravity slightly higher than that of water.

The method involves stirring 150 cc of soil sample in a 500 cc vessel containing a solution of 7 per cent aqueous magnesium sulphate (specific gravity 1.07) or 25 per cent salt solution (specific gravity 1.19) until the container is full. Many organisms will appear at the surface along with some plant debris and can be removed with a spatula into a white bottom petri dish for sorting, using a hand lens or low-power dissecting microscope. Stirring should be continued until all the large pieces of soil have disintegrated and the liquid asumes a creamy consistency. This method works well with aphids and many of the chironomidae larvae. Some tube-dwelling species and those whose bodies are coated with a layer of mud are difficult to sort out with this method. It is also not easy to separate arthropods from floating debris. They are an inherent source of error and may account for comparatively low yields of organisms such as mites and spring-tails.

MEASUREMENT TECHNIQUES FOR ORGANISMS

(a) Direct Counting

This is usually only possible in the case of stationary or slow moving organisms such as plants and animals like barnacles and molluscs. However, some animals such as large birds e.g. game birds, nesting birds or large and medium-sized mammals which occur and roam in the open can be counted directly by using a number of observers. These walk together in a line, each recording the number of animals observed in an area of ground. Care has to be taken to make allowance for those animals entering and leaving the areas while the count is in progress. To be really accurate factors such as the average rate of movement of the observers need to be taken into account but for elementary studies it provides a sound, approximation method.

(b) Indirect Counting

Many animals cannot be counted directly due to their movements and secretive habits and hence indirect methods have to be used for estimating their numbers. For some this can be done by counting their faecal pellets or droppings. Numbers can also be estimated, though with a less degree of accuracy, by counting evidence of their presence such as food remains, tracks, or burrows.

(c) Marking Methods

For estimating sizes of populations of animals over time and plotting their movements marking methods can be used. The following are some that can be adopted.

Ringing:

This is generally used for birds and mammals. It involves attaching a light metal or plastic ring with a number to a leg of the animals to be studied. Such rings can easily be constructed from a rectangular piece of 0.5 mm pure nickel foil. The required number can be embossed or scratched on the ring. In order to differentiate between different species in the same area, use of rings of different colours will prove useful.

Fur Clipping:

This is only useful for short-term studies as the fur will eventually grow back. The clipping of fur down to about 3 mm has been found to be a convenient method for marking small mammals such as rats.

Tagging:

This is mainly used for marking fishes. A commonly used tag generally consists of a short piece of stiff nickel wire bearing a metal label which is attached to the operculum, fin or tail.

Painting:

These days various types of quick drying cellulose paints have proved successful in marking land animals, in particular, those with a hard external surface, e.g. molluses and arthropods. The paint can easily be applied with a tooth pick or a sharpened match-stick. The animal should not be released until its painted mark is dry. Some paints act as an anaesthetic and hence it is advisable to ensure that marked animals are capable of dispersing on release. In the case of aquatic animals, such as gastropods, shipfouling paint may be used.

(d) Sampling

It is not always possible to count a whole population of animals or plants. The population density is therefore estimated by taking representative samples from the population concerned. These should be random as far as possible with little personal bias. The sampling method and the number and position of samples should be decided only after careful consideration of the habits and distribution of the species concerned.

Thus a preliminary qualitative study of the area under consideration proves useful before attempting quantitative analysis. This involves noting important features and places where species under study are best found. The various species in a community are not simply a random crowd. Their disposal over an area is the result of a complex of causes, and not pure chance.

In making a list of species it is desirable to record the relative importance of each species as dominant, abundant, frequent, occasional, rare or very rare. Dominant is not always the most numerous species. It is one which when removed from the area would visibly alter its appearance. Hence, the dominant species contributes most in bulk either by reason of its size or number (or both) and has the greatest physiological effect on the environment. Abundant, frequent, occasional and rare are terms used for describing relative numerical relationship.

Transects

These are lines which cut across an area of study and show graphically the changes in the composition of the fauna and flora along their respective lengths. They are positional, in contrast to random sampling, and hence great care must be taken in choosing them. A transect can be marked out on the ground with a string. Different types of transects can be devised to illustrate the general character and distribution of the fauna and flora.

Line Transect

This shows a record of fauna and/or flora occurring along a line drawn across an area. Each organism touching the line is recorded.

Profile Transect

This shows a record of plants or animals or both occurring along a line marked across an area, together with the changing level of the ground. It can also be used to show the influence of other factors, e.g. soil, water, logging, slope, etc. on their distribution.

Belt Transect

This consists of a strip across an area usually a metre wide and marked by two strings running parallel to each other.

Quadrat

A quadrat is a right-angled frame usually in the form of a square. It is used to sample an area by throwing or placing it in random situations within it. The plants and animals enclosed by each quadrat are then recorded.

Quadrats are usually made of wood or metal and hinged for folding. Alternatively they consist of string pegged out into a square. A square of one metre sides is a convenient size.

Many plants tend to grow in a clump. A square frame may cover this and miss out the surrounding species. Under such circumstances, a rectangular frame is more likely to be suitable than a square one. In order to study smaller samples, it is usual to divide the frame into smaller areas by cross-pieces either fixed or adjusted so as to slide along the main sides of the frame calibrated into centimetres.

Quadrats may also be used for estimating abundance and density of selected species. Abundance is expressed by the count of the number of individuals of each species in the quadrat. This count for each species is expressed as a percentage of the total number of individuals present in the quadrat sample. Density is simply the average number of individuals per sample over a series of samples from an area.

It is best to take samples randomly. But in view of the difficulty in obtaining truly random samples it may be more useful to increase the number of samples but reduce the size of the quadrat. The best size, however, depends on the nature of faunal and floral communities to be studied.

The profile belt transect and quadrat methods have been basically designed to study plant life. They are generally useful only for sessile or slow-moving animals.

Small and evenly distributed organisms inhabiting water or soil can be estimated by taking samples of the medium with the animals inside and counting their numbers in a given volume or area (See page 28). If enough samples are studied, a reasonably reliable estimate of the overall population density can be made.

Capture-Recapture Method

This method is useful in the case of mobile animals labelled with a ring, metal tag or spot of paint. A number of animals are caught, marked and then released. After they have had enough time to distribute themselves in a habitat, a second sample is taken and the number of recaptured marked individuals is counted. The size of the total population can now be calculated thus -

$$\frac{\text{Number in the first sample} \times \text{Number in the second sample}}{\text{Number of the marked individuals recaptured}}$$

The accuracy of the method depends on the random mixing of the marked sample before taking the second sample. It can be influenced by births, deaths, and migration into or out of the sampling area. Labelling or marking may render the animals more conspicuous to predators or hinder their movements. Birds and small mammals whose populations can be estimated by this method may become trap-shy or trap-addicted with experience and thus the proportion of marked individuals recaptured in the second sample may be misleading.

In the case of some small, active organisms population estimates cannot be made in absolute terms but the number of individuals caught in a standard manner from a locality per unit time can be compared in a relative way. However, the efficiency of the collector must be taken into consideration. This increases as the collector gains experience. Thus, several preliminary trials are necessary to achieve standardisation.

Accurate estimates of the density and distribution of animal populations are difficult to make. To overcome this problem use more than one method over a considerable period in relation to their growth and decline as well as in relation to the factors which cause these changes.

MEASUREMENTS OF ENVIRONMENTAL FACTORS

The techniques suggested here are comparatively simple, inexpensive and can be used easily and safely by students in their fieldwork. If used carefully these methods will provide results accurate enough for most school-level field studies.

(a) **Temperature**

This is an important environmental factor controlling the distribution of organisms in a habitat. It is measured by means of a thermometer. When using a thermometer, it is important to remember that the instrument should not be exposed to the direct rays of the sun and that the bulb of the thermometer should be completely immersed in the media being measured. It should be read with the bulb still in the medium after it has been in contact with it long enough for the temperature to remain constant.

An ordinary thermometer can easily be used for measuring air, soil and water surface temperatures, but its use is limited when it comes to reading temperatures at different soil and water depths. However, provided that the depth is not great, fairly accurate measurements can be made by coating the bulb of an ordinary thermometer with paraffin wax as insulator.

In the case of soil an iron tube about 5 cm in diameter can be sunk into the ground to a little more than the desired depth. About 10 - 15 cm of this tube should remain above the ground to prevent water from getting into the tube (Figure 21, page 55). A thermometer is enclosed in a glass tube of about 3 cm in diameter, which is well corked at the top and filled at the bottom with paraffin wax. The arrangement protects the thermometer and acts as an insulator when the instrument is drawn up for reading. The device is lowered and pulled through the metal tube by a chain which is attached to the cork fitted into the top of the metal tube outside the ground. A conical roof of zinc further helps to prevent rain water from getting into the tube.

In water, temperatures at different depths can be determined by using a maximum-minimum thermometer. Attach one end of the thermometer to a suitable cord calibrated in units of 10 - 25 centimetres (as required) and the other to a suitable weight. Thus the thermometer can be lowered to the required depth for any length of period and can be pulled out to record the minimum and maximum temperature reached during that time.

The simplest and least expensive method of measuring the temperature of water from depths involves the use of a "Meyer sampler bottle". This allows a moderate amount of water to be brought to the surface without undergoing any appreciable change in its temperature. Any half-litre jar or bottle can be used as a sampler. It is weighted with lead or stone (Figure 22, page 55) and is lowered with the stopper in place by means of a cliabrated cord (as mentioned above) to the desired depth. The cork is then opened by a jerk on the cord. After the bottle has been filled with water, it is pulled out and the temperature of the water sample is measured as soon as it is brought to the surface.

Electronic devices with battery power and sensing probes now available to read instantaneously the temperature of water at various depths. In using them one must ensure that their leads are well insulated. Friction against stones and vegetation tend to destroy this insulation. For continuous recording of temperatures, a thermograph can be used. It works with a simple clock within a cylindrical drum upon which temperatures are recorded in graph form by pen and ink.

Accurate temperature of microlocations such as soil, leaf litter, tree bark, wood, leaf, etc. can be recorded by resistance thermometers or thermistors and microprobes which can be buried in the material or object. The thermistors have leads projecting above the surface. To take a reading, the wires are attached to the terminals of a potentiometer, from which temperatures are read directly.

(b) Rainfall

This is an indirect factor operating mainly through soil moisture and atmospheric humidity. It can be measured with a rain-gauge consisting of a circular copper vessel, the inside of which is formed into a funnel. The latter drains rainwater into a graduated glass cylinder inside the copper vessel (Figure 23, page 55). Measurements are expressed as inches or centimetres per day or week. The apparatus is normally sunk into the ground so that its funnel is flush with the surface to reduce water loss through evaporation.

(c) Humidity

This effects the rate of water loss from both animals and plants and determines their distribution in various habitats. Direct readings of the humidity of the air can be made with a wet and dry bulb thermometer (Figure 24, page 56). The instrument contains two identical thermometers set up side by side on a suitable frame. The bulb of one is covered with a piece of muslin cloth (any absorbent material will do) connected with a small water container, while the other bulb is left exposed. The apparatus can be left in a place not exposed to direct rays of the sun and temperatures on both thermometers are read after the instrument has become adjusted to the environment. The difference between the two temperatures can then be used to determine the relative humidity with reference to Table 1, page 26.

A more efficient method for measuring humidity is a whirling hygrometer. Basically, this consists of wet and dry bulb thermometers fixed in a frame which can be rotated like a rattle (Figure 25, page 56). The hygrometer is whirled until the two temperature readings are constant. The relative humidity is then calculated as above.

TABLE 1

CONVERSION OF READINGS OF WET AND DRY BULB THERMOMETER INTO RELATIVE HUMIDITIES

Temp. °C	Depression of wet bulb thermometer in degrees Centigrade									
	1.0	2.0	3.0	4.0	5.0	6.0	7.0	8.0	9.0	10.0
0	81	64	46	29	13					
+3	84	69	54	40	25	12				
+6	87	73	60	47	35	23	11			
+9	88	76	65	53	42	32	22	12		
+12	89	78	68	58	48	38	30	21	12	4
+15	90	80	71	62	53	44	36	28	20	12
+18	90	82	73	65	57	49	42	35	27	20
+21	91	83	75	67	60	53	46	39	32	26
+24	92	85	77	70	63	56	49	43	37	31
+27	93	86	79	72	65	59	53	47	41	36
+30	93	86	79	73	67	61	55	50	44	39

Another method suitable for microhabitats involves the use of cobalt paper (strips of a filter paper dipped in 25 per cent aqueous solution of cobalt chloride ($COCl_2 6H_2O$)) and dried. There is a close relationship between the colour and the atmospheric moisture; the paper being blue at low and pale red at high humidities with a series of lilac colours in between. The length of exposure required varies with the amount of moisture and temperature but about two hours may normally be sufficient. The accuracy of this method is said to be 2 per cent when the relative humidity ranges from 40 to 70 per cent. Beyond this range the error increases to approximately 5 per cent.

(d) **Evaporation**

In some ecological studies, the natural rate of evaporation is more significant than the relative humidity. This is measured by an atmometer. Many forms of this instrument are now available. Basically it consists of an unglazed porous procelain bulb or cylinder. It is connected to a capillary tube and a reservoir. A second capillary tube containing an air bubble and fitted against a suitable scale is joined to the base of the reservoir.

The whole apparatus is filled with distilled water (Figure 24, page 56). The spherical bulb is uniformly affected by the movements of the sun across the sky and is useful for long, continuous reading. The cylinder, on the other hand, is less bulky and hence more convenient for short readings. The apparatus should preferably be used in the shade and sufficient time must be allowed for a temporary equilibrium (shown by the bubble movement coming to a steady rate) before taking the reading.

Evaporation from the porous pot results in replacement of water from below and this causes the movement of the bubble in the second capillary tube. This is timed over a given distance and a number of readings are taken to obtain a mean which can be taken as proportional to the rate of water-loss. After each measurement the moving column is refilled by turning the tap of the reservoir.

(e) Light

Sun light is the primary source of energy. It is captured by green plants through photosynthesis to power all living systems. It affects productivity (the amount of living matter that the water or land can support) and distribution of organisms.

The simplest and most accurate method to measure light intensity is by means of a photo-voltaic light meter. An ordinary photographic light meter can also be used but it has to be calibrated if the readings are required in standard units (foot-candles). It is useful for making comparative measurements in different habitats.

Often times, indirect readings using light reflected from a plain white surface are better than direct readings. However, the value recorded should be an average of several readings.

For comparisons of illumination and turbidity in shallow water, a secchi disc can be used. It can easily be made by painting a metal or plastic disc of 20 centimetres in diameter, with black and white enamel paints that are pure and striking (Figure 26, page 56). Alternatively, a white plastic dinner plate of similar diameter can be used. The disc is attached (painted face upwards) at right angles to the end of a rod of cord calibrated in units of 10 centimetres. A suitable weight is tied to the other side of the disc so that it can sink slowly when lowered into the water. To measure depth of light penetration, lower the disc in the water until it just disappears from view and record the depth. Then lift the disc slowly and note the depth at which it reappears. The average of the two readings will give the limit of light penetration into the water.

WATER SAMPLING AND ANALYSIS

Water quality influences all living systems and controls the distribution of organisms. It depends on many factors which, for the sake of convenience may be grouped into three main categories: physical, chemical and biological. Physical factors include temperature, light penetration, turbidity and amount of suspended particles. Biological factors involve kinds and numbers of living organisms present. Chemical factors include pH, hardness, amounts of dissolved gases, phosphates, nitrates, etc. All these factors interact with each other to influence the quality of water. This section deals mainly with some selected chemical factors and the respective techniques for measuring them.

Collecting Water Samples

The sampling of water from the surface down to arm's-length depth generally presents no difficulty. It can be achieved by lowering a screw-top bottle or jar in the water, removing the cap under water and allowing the bottle to fill on its own before pulling out the water sample with the lid tightly secured. In replacing the stopper, care must be taken to ensure that no air bubbles are trapped in the bottle.

For deeper samples a heavy glass bottle (200 ml. or 250 cc) fitted with an air-tight ground-glass stopper is used. This is weighted with lead or a stone heavy enough to sink it when full of air. The stopper is loosely inserted into the bottle which is then lowered gently into the water by means of a string calibrated in units of 10 centimetres. On reaching the required depth, the stopper is removed by jerking the string to allow the water to enter the bottle. When it is full, it can be pulled out. With such a method, a little air is likely to dissolve in the water before the stopper can be replaced.

An alternative method for the purpose has been described by McLean and Cook (1968). The apparatus involved is shown in Figure 28 page 57. It consists of a bottle (250 cc) fitted with a rubber stopper and weighted by thin lead piping wound round its bottom. The double right-angled glass tube on the left serves as the water inlet and passes through the rubber stopper to the bottom of the bottle. The tube on the right fits flush with the rubber bung to avoid trapping air bubbles and passes into a second smaller bottle (c). This is to receive the overflow of water from the larger bottle.

The whole assesembly is secured in a light wooden frame and slung from a central metal ring (d). The rubber tube 'b' is blocked with a stopper (a piece of glass rod) which can be pulled out by an independent string. As long as this remains intact, water cannot enter the sampler. On pulling the stopper at the required depth, water rushes in to fill both the bottles. After taking the water sample, the apparatus can be drawn out; the water in the small bottle is rejected while the rubber bung of the larger bottle is carefully removed and replaced by a ground glass stopper. The water in the large bottle provides the sample for analysis. However, special water samplers are

available that make it possible to secure water without contamination with atmospheric oxygen. However, in transferring water from the sampler into a reagent bottle, the latter should be allowed to overflow two or three times its capacity to eliminate contamination from atmospheric oxygen.

All water samples should be carefully labelled indicating the date, time and location soon after their collection.

Water Analysis

In addition to dissolved gases, natural water may contain three kinds of solids - sediments, suspended solids and dissolved solids. Sediments are those particles which settle down when the water is allowed to stand. The truly suspended material, on the other hand, does not settle out. Dissolved solids refers to minerals and salts which exist in solution.

(a) Determination of Sediments in Running Water:

Stir the water sample in a convenient container until no material is visible at the bottom.

Quickly pour this stirred sample into a graduated cylinder up to the top mark and allow it to stand for 24 hours.

Measure the volume of the material that has settled down in the cylinder and express it in terms of percentage of the total sample in the graduated cylinder.

In order to determine the weight of the sediments in water, slowly decant off the top liquid in the cylinder without disturbing the sediments at the bottom. Weigh a clean dry beaker or crucible, transfer the sediments into it, rinsing the material from the cylinder with distilled water.

Gently heat the material over a water bath (without boiling or spluttering) until all water evaporates. Cool in a dessicator and weigh the residue with the container and determine the weight of the sediments by subtracting the weight of the container from the total.

Express the result in grams of sediments per ml. of water by dividing the net weight of the residue by the volume of water originally placed in the graduated cylinder.

(b) Determination of Suspended Solids in Water:

Such materials may include very fine particles of clay, organic debris, waste materials in sewage or industrial discharges and plankton; and their presence in the water is determined by filtering the water sample.

For better results, the Millipore Sterifil Filtration System can be used (Figure 29, page 57). Assemble the system by pressing the filter base firmly down over the receiver flask so that the filter support should be in place at the top of the filter base. Place a filter on the filter support (grid side up) by means of forceps and carefully centre it on the filter support. Screw the 250 ml. sterifil funnel on the filter base, making sure that the filter remains centred. Attach the vacuum syringe to the long part of the receiver flask and place a rubber cap over the short part on the opposite side of the flask.

Pour a known volume of the water (using a graduated cylinder) into the sterifil funnel. If the sample is relatively clear use 250 ml., otherwise a smaller amount will do.

Apply vacuum to the system by working the plunger of the vacuum syringe up and down. Thus the air will be drawn out of the receiver and its place will be taken by the liquid pushed through the filter by the atmospheric pressure. Any suspended solid material present in the sample will be trapped on the filter. If the sample is rich in suspended material, the filter is likely to become clogged. This will either stop the passage of water or allow it to go through only very slowly - drop by drop. If this happens, you may have to repeat the experiment using a smaller sample volume. After all the liquid has passed through the filter disconnect the vacuum syringe from the part of the receiver flask. Unscrew the filter funnel and using the forceps transfer the filter from its support to a clean paper towel to dry. The filtered water may be saved for analysis of dissolved solids.

The dried filters from various samples can be mounted on a clean sheet of white paper for comparison. The difference in intensity of colour of the filters provides a visual indication of the relative amounts of suspended materials collected.

If a good analytical balance is available, gravimetric analysis can be undertaken as follows:

(a) weigh a clean, dry Millipore filter accurately (W1);

(b) filter the water sample as stated above;

(c) allow the filter with the suspended material to dry completely;

(d) reweigh the filter as dried in (c) (W2);

(e) determine the weight of suspended solids (W3) = W2 - W1

The concentration of the suspended solids per litre of water sample =

Weight of suspended solids (W3) in milligrammes X 1000.

(c) **Determination of Dissolved Solids:**

A rough estimate of total dissolved solids can be made by taking 100 ml. of the sample in a dry weighted crucible (W1) and evaporating it gently over a water bath, taking care not to allow spluttering or to burn the residue. The crucible with the residue is then cooled in a dessicator and re-weighed (W2). The difference in weights (W2 - W1) gives the total amount of dissolved solids in 100 ml. of water. This can be converted to the weight of total dissolved solids per litre as follows:

$$\frac{W2 - W1}{100} \times 1000 = \text{Total dissolved solids in mg. per litre of water sample}$$

For more accurate estimates the following procedure can be used. This is performed using the sample from which sediments and suspended material have been removed.

Thoroughly wash a 250 ml. beaker and a 100 ml. graduated cylinder with soap and water and rinse them with tap water followed by distilled water. Filter the water sample using the Millipore Sterifil Filtration System as explained above. If this apparatus is not available, the water sample can be filtered through ordinary filter paper using a normal filter funnel.

Measure 100 ml. of the duly filtered water sample with the graduated cylinder and evaporate it gently in a weighted beaker (W1) over a water bath as mentioned earlier or in an oven at 80°C. Weigh the dry beaker with the residue (W2). The weight of the dissolved solids can be found:

W2 - W1 = weight of dissolved solids per 100 ml. of water sample (W3)

The concentration can then be determined:

$$\frac{\text{Weight of dissolved solids in milligrammes}}{} \times 1000 = \text{mg. dissolved solids per litre of water sample}$$

(d) **Salinity (Chlorides) Measurements:**

Salinity of sea water varies little, but in estuaries its fluctuations may be considerable. It can be determined by titrating 100 cc. of the water sample with N/100 (0.01N) silver nitrate solution using a few drops of potassium chromate solution as indicator. The end point is reached when a faint red colour of silver chromate appears. Multiply the number of cubic centimetres of 0.01N silver nitrate used by 0.000355 to obtain the chlorides in parts per million.

(e) **Total Hardness of Water**

The hardness of water is due to salts of calcium and magnesium present in the water. The degree of hardness is expressed in terms of parts per million (pp m) of calcium carbonate present in the water. Thus soft water has a hardness of 0 - 60 ppm; moderately hard water, 61 - 210 ppm; hard water 121 - 180 ppm; and very hard water, over 180 ppm. Hardness values below 250 ppm are considered acceptable for drinking and values above 500 ppm as hazardous to human health.

Temporary hardness is due to the bicarbonates of calcium and magnesium and can be estimated in the same way as combined carbon dioxide in neutral and alkaline waters (See page 35)

Permanent hardness is caused by the chlorides and sulphates of calcium and magnesium. It can be determind as follows:

Add 50 cc N/10 sodium carbonate solution to a 250 cc sample of water in a flask or beaker and boil for at least half an hour in order to convert any bicarbonate present into insoluble carbonate.

Filter the contents into a 250 cc volumetric flask washing well the precipitated matter with a little hot distilled water. Allow the filtrate to cool and make it up to the required volume of 250 cc with distilled water, shaking well.

Take 50 cc of the filtrate and titrate it against N/10 sulphuric acid using methyl orange as an indicator.

1 cc of N/10 sodium carbonate = 0.005 g. calcium carbonate

50 cc of filtrate required X cc of N/10 sulphuric acid

$\therefore$ 250 cc of filtrate would require 5 X cc of N/10 sulphuric acid

But 50 cc N/10 sodium carbonate were added originally

$\therefore$ 50 - 5 X cc sodium carbonate were used

Hence hardness = $\frac{(50 - X) \times 0.005 \times 100{,}000}{250}$ ppm

(f) **Determination of Dissolved Oxygen**

The Winkler method, though cumbersome, provides reliable and accurate results. The following reagents are required:

(A) 40 per cent Manganous chloride solution

(B) 33 gms. of sodium hydroxide and 10 gms. of Potassium iodide dissolved in 100 cc of distilled water

(C) N/80 Sodium thiosulphate solution. This solution should be standardised in the usual way against Potassium dichromate or Potassium permanganate. The solution does not keep well for long and any appearance of a precipitate is a sure sign of its deterioration. Though this can be delayed by the addition of 5 cc Chlorotorm or 3 gms. of borax per litre, it is advisable to prepare the solution not too long before the field work.

(D) Concentrated Hydrochloric acid or Ortho-phosophoric acid.

(E) Starch solution as an indicator. This can be prepared by boiling some starch in distilled water. The solution must be cooled before use.

Procedure

Collect the water sample as described earlier using a 70 cc bottle. The bottle should be well-stoppered and completely filled with water so that it has as little contact with air as possible.

Remove the stopper and add 0.5 cc of solution A and 0.1 cc of solution B ensuring that no air bubbles are included. For volumes greater or less than 70 cc calculations of amounts of solutions A and B to be used must be made accordingly.

Leave to stand for five minutes when a brown precipitate of Manganous hydroxide will appear indicating that oxygen of the sample has been fixed. This step is best carried out in the field and the rest in the laboratory.

Add about 2 cc of reagent D. After carefully replacing the stopper mix by rapidly inverting and rotating the bottle until all the precipitate is dissolved and the colour of the liquid changes to a clear golden brown due to the liberation of iodine.

Transfer 25 cc of the above liquid with a pipette into a small flask and titrate it against solution C using a drop or two of solution E as an indicator until all the golden brown colour of iodine has disappeared.

The procedure should be repeated at least twice for each sample.

Calculations

1 cc of N/80 sodium thiosulphate is equivalent to 0.1 mg of oxygen and if 'x' is the volume of thiosulphate solution used and 'y' the volume of the water sample then

$$\text{the amount of oxygen/litre} = \frac{X \times 0.1 \times 1000}{y} \text{ mg}$$

Now 1 cc of N/80 Sodium thiosulphate = 0.0011 g of oxygen

1 cc of N/80 sodium thiosulphate =

$$\frac{0.0001 \times 22{,}400}{32} \text{ cc of oxygen}$$

Hence $\frac{X \times .00001 \times 22{,}400 \times 1000}{32 \times y} = \frac{X.\ 70}{y}$ cc of oxygen/litre at N.T.P.

This method follows one described by Dowdeswell (1967). Sources of error could be the presence of nitrite - a rare occurrence in natural water. If present it must first be oxidised to nitrate with potassium permanganate; the excess permanganate if any can be neutralised with Potassium oxalate solution. Some static waters may contain appreciable quantities or organic matter. This must be removed if accurate results are to be obtained. An easy way to do so is to oxidise the organic substance with bromine and to remove the excess by addition of sodium salicylate. The following reagents are required for the purpose:

(A) 3 gms. of Potassium bromate, 20 gms. of sodium bromide and 25 cc of concentrated Hydrochloric acid made up to 100 cc with distilled water

(B) 10 gms. of salicylic acid and 20 cc of 15 per cent caustic soda made up to 100 cc with distilled water.

Procedure

To 100 cc of the water sample add 0.5 cc of solution A. Re-stop and immerse the container in water in the dark for 24 hours. Then add into it 0.5 cc of solution B and shake well. Allow it to stand for 15 minutes. The subsequent procedure is as for the Winkler test described earlier.

(g) Estimation of Carbon Dioxide

It is advisable to determine the pH (see page 36) of the water sample before proceeding to estimate the carbon dioxide present in it.

(i) Acid Waters - free carbon dioxide

Such waters contain free carbon dioxide which can be determined by using the following reagents:

(A) N/100 sodium carbonate solution. This is prepared by dissolving 0.53 g of pure anhydrous sodium carbonate (dried in an oven at 103° C for an hour) in one litre of distilled water. Since the solution does not keep well, it should be freshly prepared and standardised before use against N/100 Hydrochloric acid.

(B) One per cent phenolpthalein solution in 50% alcohol neutralised with dilute sodium hydroxide.

Procedure

Add 1 cc (about 10 drops) of phenolphthalein solution to 100 cc of water sample in a measuring cylinder or a similar container which presents a minimum surface of the water to the air in order to avoid the formation of bicarbonate by contact with air.

Titrate it with solution A (sodium carbonate) until a persistent (at least for 3 - 4 minutes) pink colour is obtained. While titrating, the container should be stirred only gently to further minimise error indicated above.

Calculations

If X = the number of cc of N/100 sodium carbonate required by 100 cc sample of water, then the normality of CO_2 = $\frac{X}{10,000}$

Since the normal solution of CO_2 contains 22 g per litre

the sample contains 2.2 X mg. per litre

i.e. 2.2 x parts per million.

By multiplying this figure by 0.509, the result can be expressed as cc per litre at N.T.P.

(ii) **Neutral and Alkaline Waters - Combined Carbon Dioxide**

In such waters carbon dioxide is estimated as bicarbonate - mainly that of calcium and magnesium. Reagents required are:

(A) N/100 Hydrochloric acid. This can be standardised against firstly prepared sodium carbonate solution (as described in the test of acid waters).

(B) Methyl orange (1 - 1,000) in distilled water as indicator.

Procedure

Take 100 cc of the water sample. Add a few drops of the indicator (B) and titrate against N/100 Hydrochloric acid (A) as stated in the case of acid water sample. The amount of combined carbon dioxide is expressed as the equivalent quantity of calcium carbonate per litre. Let X = the number of cc of N/10 HCL required for 100 cc of water sample. The equivalent of Ca CO_3 is 50. Hence the amount of CO_2 stated as $CaCO_3$ will be 5 X mg. per litre (5X parts per million).

(h) Determination of pH (Hydrogen-ion concentration)

Individual indicators can be used for the purpose. They change colour at known pH values as indicated in the table below:

Name of Indicator	pH	Colour Change
Bromophenol blue	2.8 - 4.6	yellow to violet
Bromocresol green	3.6 - 5.2	yellow to violet
Methyl red	4.4 - 6.2	red to yellow
Bromocresol purple	5.2 - 6.8	yellow to violet
Bromothymol blue	6.0 - 7.2	yellow to blue
Phenol red	6.8 - 8.4	yellow to red
Cresol red	7.2 - 8.8	yellow to violet-red
Thymol blue	8.0 - 9.6	yellow to blue
B.D.H. "soil" indicator	4.0 - 8.0	red through green to blue
B.D.H. "4 - 11" indicator	4.0 - 11.0	red through green to violet
B.D.H. "Universal" indicator	3.0 - 11.0	red through green to violet

Special colour charts for the purpose are available. More accurate estimation can be made by using a capillator with single indicator (See Soil Analysis, page 42)

SOIL ANALYSIS

Soil is a vital factor in terrestrial habitats including agriculture. Before undertaking any ecological study of soil it is useful to know something of the geology of the area from an office of the geological survey in the district.

Soil Sampling

The usual methods for extracting soil samples include:

Digging: Approximately assessed soil samples can be obtained by digging using a sharp knife, a trowel or, if large samples are required, a spade.

Using an Apple-corer type sampler: Basically such a sampler consists of a steel cylinder graduated on the exterior at 5 cm. intervals to enable samples to be taken at a required depth. The cylinder is 25 - 30 cm. in length and about 5 cm. in diameter. One of its ends has a sharpened rim which can be driven into the soil by means of a handle provided at the other end. A slit on the cylinder can help to force out the required length of the sample (Figure 30, page 57)

Using a soil auger: The auger is like a large screw with a handle on top. . It is suitable for sampling heavy soils. It can be screwed into the soil to any depth and is then pulled out directly. The soil collected in the thread of the auger can be removed for study.

Soil Profile

This is a vertical, side-view of the soil constituting a summary of the properties of the soil. In studying a profile a scaled sketch should be made and full description of each layer or horizon should be noted as follows:

(a) Name of the horizon along with its depth, indicating clearly its boundaries. Thus, details of layers such as litter, humus, etc. should be noted.

(b) Colour whether uniform or otherwise. It is usually due to humus (brown, grey or black), iron compounds (red, orange or yellow), and calcium (white).

(c) Texture and constitution indicating whether sandy, clayey, loamy, peaty, etc.

(d) Proportion and nature of stones present.

(e) Organic matter indicating its nature and distribution with special reference to structural remains of plants.

(f) Root development providing clues for moisture and aeration.

(g) Moisture and drainage.

Mechanical Analysis

Soil particle-size is significant in determining the ecological characteristics of a soil and is also used as a criterion in classifying most of its solid constituents. Several classifications are in use. For details on the subject reference should be made to some standard textbooks on soil and agriculture. The following classification, however, is simple and convenient for our purposes.

Clay	Less than			0.002	mm.
Silt		0.002	-	0.02	mm.
Fine sand		0.02	-	0.2	mm.
Coarse sand		0.2	-	2.0	mm.
Stone (gravel)	More than			2.0	mm.

The relative proportion of each grain-size group can be determined by sieving to remove stone and sand and then separating the silt from the clay by differential settlement in water. The following method is simple and convenient for our purposes.

(i) Take a weighed sample of oven-dried soil. Rub it gently in a mortar with a wooden pestle to break the clumps and pass it through a nest of sieves to separate stone, coarse sand and fine sand and weigh the separated fractions.

(ii) Collect the particles (silt + clay) passing through the fine sieve in a large weighed evaporating dish (W1) and re-weigh the dish (W2). Determine the weight of silt and clay by subtraction i.e. W2 - W1 = W3.

(iii) Add distilled water so that its level stands about 1 cm. above the fine material in the re-weighed dish.

(iv) Stir the mixture well, adding more water if needed and allow it to stand for several hours and decant off the cloudy supernatant liquid.

(v) Repeat the above process until the supernatant liquid is clear (3 - 4 stirrings may be enough).

(vi) Evaporate to dryness, and weigh the silt remaining in the evaporating dish (W4).

(vii) Determine the weight of clay fraction in the decanted liquid by subtracting W4 in (vi) from W3 in (ii).

(viii) Represent respective weights of different soil particle-size fractions as percentages of the weight of the oven-dried soil sample under (i) for comparison.

Water Content

Soil moisture is a significant influence on the distribution of soil fauna but its direct measurement is said to have only little value as far as plant ecology and agriculture are concerned. It is useful to determine available water and field capacity along with the total water present in the soil for better understanding of the role of water in the soil. Sampling for the purpose should be avoided when the soil is saturated due to heavy rainfall. It is suggested that a period of at least 24 hours should be allowed for the excess water to drain away.

(a) Amount of Total Water in Soil

(i) Take some soil and weigh it carefully and note the weight (W1).

(ii) Heat it to a constant weight at just over 100°C in an oven so that all the uncombined water is driven off. Care should be taken that the organic water is not destroyed.

(iii) After cooling in a desiccator, re-weigh the dried sample and note the weight (W2).

(iv) Subtract W2 from W1 to obtain the weight of the water loss (W2 - W1 = W3).

(v) Express the result as percentage of the total weight of the soil sample, i.e. W1.

$$\text{Percentage of total water in soil} = \frac{W3}{W1} \times 100.$$

It must be understood that single estimates mean very little and hence a series of samples taken from different places at the same time should be analysed to obtain reliable comparison.

(b) **Available Water**

The amount of available water in a soil sample is determined in a way similar to one described above under (a) 'Total Water in Soil'. However, for this purpose, the sample is air dried to a constant weight at room temperature.

(c) **Field Capacity**

This refers to the maximum amount of water which a freely drained soil can hold. In estimating this the soil sample should be used in its natural condition i.e. neither broken nor compressed.

(i) Take a soil sample using a tall, slim can. Cut off the roll-edged end and perforate the other end. Weigh this "can sampler" (W1). Drive it to the ground until the performated end is flush with the surface.

(ii) Carefully dig the can out of the ground with the contained soil sample, wipe its outer surface clean and weigh (W2).

(iii) Lower the can into water, perforated end first, and leave it until the soil is thoroughly soaked.

(iv) Remove the can from water and allow it to drain freely.

(v) When water stops dripping, dry the outside of the can carefully with blotting paper and weigh (W3). Any increase in weight (W3 - W2) is due to water uptake and may be expressed as percentage of the original soil sample weight (W2 - W1) as

$$\frac{\text{Weight of soil after soaking and draining}}{\text{Original weight of soil}} \times 100 = \frac{(W3 - W2)}{(W2 - W1)} \times 100 = \text{Percentage increase of water uptake}$$

(v) mark the new water level after removing the soil as "y";

(vi) after washing out the can and jar, refill the jar with water up to the original level (x);

(vii) seal the perforations of the can with an adhesive tape or paraffin wax and after filling it with water to the brim, lower it into the jar;

(viii) mark the new level of water as "z";

(ix) determine the amount of water needed by pouring it with a measuring cylinder to raise the level from mark (y) to mark (z). This will be equivalent to the amount of soil air displaced.

Assuming 1 ml. of water weighs 1g., the amount of water represented by (z - y) can be expressed as an increase in weight. The maximum water capacity can be calculated in the same way as field capacity under (c) above.

Organic Matter

For all practical purposes, soil organic matter relates to the amount of humus in the soil. Its accurate determination is difficult but loss in weight on burning to red heat can provide a reasonable estimate of soil organic content for comparing different soils.

(i) Take a clean crucible and weigh it (W1);

(ii) Weigh an oven-dried soil sample in a crucible cool and keep in a desiccator to avoid uptake of moisture (W2);

(iii) Heat the weighed soil in (ii) to red heat for about half an hour to oxidise the organic matter;

(iv) Cool in a desiccator, weigh and reheat until the weight is constant (W3);

(v) The loss in weight (expressed as a percentage) equals a reliable estimate of the oxidisable organic content of the soil.

$$\text{Percentage of organic matter} = \frac{\text{Weight of soil after burning}}{\text{Height of the oven-dried soil}} \times 100$$

$$= \frac{W2 - W3}{W2 - W1} \times 100$$

One of the chief sources of error in this method is the decomposition of organic minerals particularly carbonates.

Thus if the increase after soaking and draining is 20 per cent of the original soil weight, 100 g. of soil will take up 20 g. of water.

(vi) To estimate the field capacity, oven-dry the sample with the can after the water has stopped dripping and weigh (W4).

$$\text{Feild capacity} = \frac{\text{Weight of the soil after soaking and draining} - \text{weight of the oven-dried soil}}{\text{Weight of the soil after soaking and draining}} \times 100$$

$$= \frac{(W3 - W1) - (W4 - W1)}{(W3 - W1)} \times 100$$

Suppose that:

Weight of the original soil sample	=	100 g.
Weight of the soil after soaking and draining water	=	120 g.
Weight of the oven-dried sample	=	60 g.
Amount of water	=	120 - 60 g.

Thus, the soil sample after soaking and draining weighs 120 g. and contains 60 g. of water. Hence it has a field capacity of

$$\frac{60}{120} \times 100 = \text{i.e. } 50\%$$

(d) **Maximum Water Capacity**

This refers to the maximum amount of water the soil can hold if drainage is prevented and all air displaced. In terms of volume it is equivalent to pore space in the soil and can be determined as follows:

(i) collect a soil sample using a weighed tin can (W1) as described under (c);

(ii) level off the soil with the open end of the can and weigh it (W2);

(iii) take a wide gas jar or a similar container and fill it with water to a level which will just cover the can and mark this level as "x";

(iv) lower the can with the soil sample into the water in the jar and carefully remove the soil to displace the air;

This can be corrected by adding a little ammonium carbonate solution to the cooled solid and then heating on a water bath or in an oven at about 105°C. to drive off the excess. Cool and reweigh. The gain in weight will represent the amount of carbon dioxide lost by the carbonates during the initial heating.

The crucible should be used with the lid to avoid "spitting" during heating.

pH (Hydrogen-ion Concentration)

Soil acidity and alkalinity is measured on the pH scale. The methods of estimating pH of water already described (page 36) are equally applicable to the study of soil. Most of these methods are based on colour changes in indicator solutions. Some of these are described below:

(i) Place about 2 g. of soil on a crucible lid, soak it with BDH Universal indicator, wait 1 - 2 minutes and then tilt the lid so that indicator drains out of the soil. Compare the resultant colour of the drained liquid with the chart supplied with the Indicator and determine the pH.

(ii) Soak about 2 g. of soil on a crucible lid with distilled water. Drain the water and test it first with 'Universal Indicator Test Papers' followed by those with limited range to obtain a more accurate pH value of the soil.

(iii) BDH Soil Test Outfit. In using this a small quantity of soil is mixed with distilled water and barium sulphate in a tube and shaken. The barium sulphate ensures flocculations and precipitation of colloidal clay at the bottom of the tube leaving a clear liquid above. The colour of this liquid, after adding the indicator, can be matched against the chart of standards to determine the soil pH. A poor quality barium sulphate can give rise to inaccuracy due to its acidity.

Soil Minerals

Presence or absence of simple inorganic minerals in soil water can vitally influence the growth of plants and hence the distribution of animals. Most of the methods used for chemical analysis of water are equally applicable to soil analysis. They are described in detail in standard textbooks on agricultural and soil chemistry and should be consulted when more accurate estimates are to be made. The colorimetric tests are simple and reasonably accurate and are described here for comparative studies in elementary ecological work.

Preparation of a Soil Extract

Before attempting the mineral analysis of soil it is essential to prepare a soil extract. To do this:

(a) (i) Take a fresh sample of soil and dry it in an oven or a steam bath at about 105°C. to stop bacterial action.

(ii) Store and cool in a desiccator.

(iii) Transfer about 250 g. of the dried soil into a Buchner funnel and wash with 1 litre of hot distilled water.

(iv) Allow the filtrate to cool and make up to 1 litre in a volumetric flask.

If the resulting liquid is cloudy or coloured proceed as follows:

(b) (i) Dissolve 125 g. potassium or ammonium alum in a litre of distilled water.

(ii) Add ammonium hydroxide into the above solution until no more precipitate of alumina appears.

(iii) Wash the precipitate well and then mix the solid with distilled water making upto a litre.

(iv) Add about 1cc. of this to every 150 cc. of the soil extract (if cloudy) prepared under "a". Shake well, allow to settle and filter if necessary before testing.

Estimation of Total Inorganic Nitrogen

This is conveniently estimated as ammonia. The following reagents are required for the purpose.

(A) **Nesslers' Solution:**

Dissolve 10 g. potassium iodide in 10 cc. distilled water. Add a saturated solution of mercuric chloride until a slight permanent precipitate is formed. Again add 80 cc. of 9 M potassum hydroxide solution (504 g. per litre) and make up to 200 cc. with distilled water. Allow to stand for 24 hours and then filter.

(B) **Zinc-copper couple:**

Allow some granulated zinc to stand in dilute copper sulphate solution for a few minutes until zinc is well coated with copper. Wash and dry. Zinc-copper couple does not keep well and a fresh supply should always be prepared as needed.

(C) Concentrated hydrochloric acid free from ammonia compounds.

(D) Dilute caustic soda solution free from ammonia compounds.

Chemicals (C) and (D) can be tested with Nessler's solution to ensure that no ammonium compounds are present.

Procedure

(i) Take about 250 cc. of soil extract;

(ii) Filter to remove all suspended matter (best done by Buchner funnel and filter pump);

(iii) Take two 100 cc. samples of (ii) and to each add about 5 g. zinc-copper couple and 5 drops of concentrated hydrochloric acid and stir well;

(iv) Incubate at 35 - 40°C. for 6 hours to complete the reduction of the nitrogen present;

(v) Make the liquid alkaline by adding a small amount of caustic soda solution and distil the resultant for at least 15 minutes.

(vi) Collect the distillate in about 100 cc. of cold distilled water.

(vii) Test a few drops of the above liquid with Nesslers' reagent. Absence of orange yellow colour indicates the removal of all ammonia.

(viii) Make up the liquid in (vi) to a convenient volume say, 200 cc. transfer 45 cc. of this to a 50 cc. Nessler Tube and add 5 cc. of Nesslers' solution. A clear orange-brown colour indicates the presence of ammonia. The formation of brown precipitate indicates that considerable amounts of nitrogen are present and the solution must be diluted accordingly in order to obtain accurate results.

To make a standard for comparison, dissolve 3.82 g. of dry freshly sublimed ammonium chloride in a litre, of distilled water. This constitutes a stock solution containing 1000 parts of nitrogen per million. It can be diluted and tested as in (viii) for colour comparison. This can be done by standing the Nessler tubes on a white surface and looking on to the surface of the liquid. For quick comparison a lovibond comparator (or similar colorimeter) comprising a series of coloured discs corresponding to the tints produced in Nesslers' solution by different concentrations of ammonia can be used.

By omitting the reduction with zinc-copper couple, the method can also be used for estimating soil ammonia only.

The following precaution should be observed in using the nitrogen analysis method:

(i) Nessler's solution should always be added slowly and in cold. Warming produces a yellowish precipitate and no colour comparison is then possible.

(ii) A similar precipitate is formed when certain ions espeically chlorine is present. It is essential, therefore, to collect the distillate in water and not in hydrochloric acid.

(iii) All glassware must be scrupulously clean. Glass stoppers should be used in the distillation apparatus. If corks are used, they must be covered with tinfoil.

(iv) Two similar samples should be tested together as a check on accuracy.

(v) The orange-yellow liquid formed in the Nessler reaction does not keep well more than a day. A fresh series of standards should therefore be prepared on each occasion from the various stock solutions of ammonium chloride, which can keep indefinitely.

(vi) If an addition of Nesslers' solution produces a cloudiness the following can prove as an effective remedy:

Dissolve 50 g. sodium potassium tartrate in 100 cc. distilled water and filter. Now add 5 cc. Nessler's solution to the filterate, allow it to stand for a few days and filter off any brown precipitate. Add an equal amount of this solution to Nessler's reagent just before use. The resulting fluid is unstable and must be prepared afresh as required.

Estimation of Nitrate

The Sprengel method has been found to be quick and reliable for this purpose provided the amount of chloride present in the soil does not exceed 30 parts per million. This difficulty may only arise when dealing with marine or estuarine soil in which case total chloride present must first be estimated and neutralised as follows:

Add dilute sulphuric acid to a convenient volume of soil extract or water until neutralisation is complete. Then treat with an appropriate quantity of standardised silver sulphate solution (prepared by dissolving 4.397 g. of pure, dry silver sulphate in 1 litre of distilled water, 1 cc. $\equiv$ 0.001 g. chloride). This will precipitate all but a trace (about 10 parts per million) of the chloride present. Filter well.

Reagents needs for the estimation of nitrate include:

(A) **Phenol-sulphuric acid:** If the ready made chemical is not available, proceed as follows: add 18 g. of pure phenol to 9 cc. of distilled water in a 250 cc. evaporating dish. Then pour 111 cc. of pure sulphuric acid slowly in the evaporating dish and heat on a water bath at 100°C. for about 8 hours. This provides the stock solution which will keep well in the dark.

(B) **Potassium or Sodium hydroxide solution:** 240 g. caustic soda dissolved in 500 cc. of distilled water.

(C) **Potassium nitrate:** Used as a standard, dissolve 0.722 g. pure potassium nitrate in a 1 litre with distilled water. Dilute 100 cc. of this solution to 1 litre with distilled water to obtain a concentration of 10 parts of nitrate per million (i.e. 1 cc. of solution = 0.00001 g. nitric nitrogen).

(D) **Aluminium sulphate:** 10% solution in water.

(E) **Concentrated ammonia.**

Procedure

(i) Take 10 cc. of soil extract (more if the amount of nitrate present proves to be small) and evaporate on a water bath until dry. Do not heat on a flame.

(ii) Allow to cool and add 1 cc. of phenol sulphuric acid and 1 cc. of distilled water.

(iii) Mix well and warm on a water bath for 10 minutes and add about 2 cc. aluminium sulphate solution and just enough ammonia to make the liquid alkaline.

(iv) Dilute the above solution with distilled water to 100 cc.

(v) Transfer 50 cc. of above to a Nessler Tube and compare the yellow colour with that obtained by treating 10 cc. of standard solution in the same way.

The above method seems most accurate when the nitrate concentrations are between 1 and 10 parts per million. Beyond this limit colour match is difficult and hence appropriate dilution of the soil extract is desirable before testing.

The standard coloured solutions do not keep well over a few hours and hence must be prepared afresh for each testing. As in the case of total nitrogen, the Lovibond comparator, if available, can be used for greater speed. The standard coloured discs should be matched against actual standards prepared in the laboratory from time to time.

Estimation of nitrites

They are found in soils containing much decaying matter. The Griess-Ilosva method can be used successfully for their estimation. It needs the following reagents;

(A) **Sulphuric acid solution:** Dissolve one g. of pure sulphuric acid in 60 cc. glacial acetic acid and 240 cc. distilled water.

(B) **Napthylamine solution:** Dissolve 0.2 g. of Naphthylamine in 60 cc. of glacial acetic acid and 240 cc. distilled water.

(C) **Stock nitrite solution (standard):** Dissolve 1.1 g. silver nitrite in distilled water and precipitate the silver with sodium chloride. Dilute to one litre. Again dilute 5 cc. of this solution to one litre. Then add 1 cc. of chloroform as a preservative. The concentration of the solution is now 0.5 mg. nitrogen per litre. Thus 1 cc. of this solution = 0.0005 mg.; N = 0.00164 mg. nitrite.

Procedure

(i) Take 100 cc. of soil extract in a Nessler tube and add 1 cc. of each of the solutions (A) and (B).

(ii) Allow to stand for 10 minutes; appearance of a pink colour shows that nitrite is present.

(iii) Match the resultant colour with tints derived from different concentrations of the standard nitrite solution which has been treated in the same way.

Since the colour deepens on standing, it is important that the time allowed after the addition of reagents should always be the same. A fresh set of standards must be prepared for each test.

Estimation of Phosphates

These usually occur in small amounts, not exceeding 1 part per million of water but they are very important minerals influencing plant and animal growth. The molybdenum blue test is convenient for their colorimetric estimation. This requires the following chemicals.

(A) **Standard phosphate solution:** Weigh 0.4263 g. pure ammonium monohydrogen phosphate [(NH4)2 HPO4] and make it up to a litre with distilled water (1cc. $\equiv$ 0.1 mg. phosphorus). Add one crystal of potassium permanganate as a preservative. Dilute 100 cc. of this solution to 1 litre (1 part of phosphorus per million) to make stock solution.

(B) **Molybdate - sulphuric acid reagent:** Mix 100 cc. of 10 per cent ammonium molybdate solution with 300 cc. of 50 per cent (by volume) sulphuric acid. This should be stored in a dark bottle as the reagent can decompose rapidly in the light.

(C) Prepare 2.5 per cent stannous chloride solution by dissolving 2.5 g. of the substance in 10 cc. of concentrated hydrochloric acid. Make it upto 100cc. with distilled water, filter and store in small stoppered bottles each containing a piece of pure tin.

Procedure

(i) Transfer 100 cc. of soil sample extract into a Nessler tube and add 1 cc. of solution (B) followed by about 5 drops of solution (C).

(ii) Mix well and allow to stand for 10 minutes.

A blue colour will appear indicating the presence of phosphorus. This should be matched against comparable standards made up from solution (A) and treated the same way. The result can be expressed as parts of phosphorus per million. "Molybdenum" blue is unstable.

Carbonate content

This is mostly calcium carbonate. A rough estimate can be obtained as follows:

(i) Take about 5 g. of the soil sample in a tube. Add some water and boil to remove air bubbles.

(ii) Add 10 ml. of dilute hydrochloric acid; effervescence will result. Slight effervescence indicates that less than 0.5% calcium carbonate is present while strong effervescence indicates an amount exceeding more than 1%.

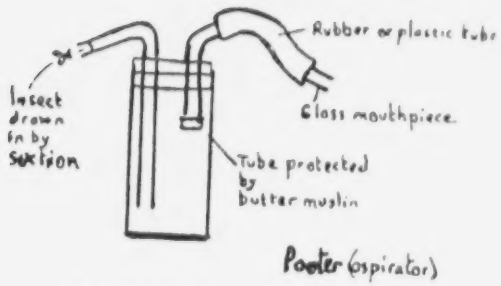


FIGURE 3

FIGURE 4

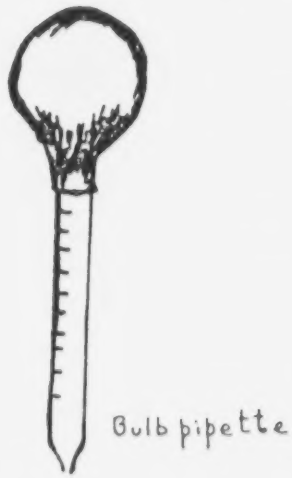


FIGURE 5

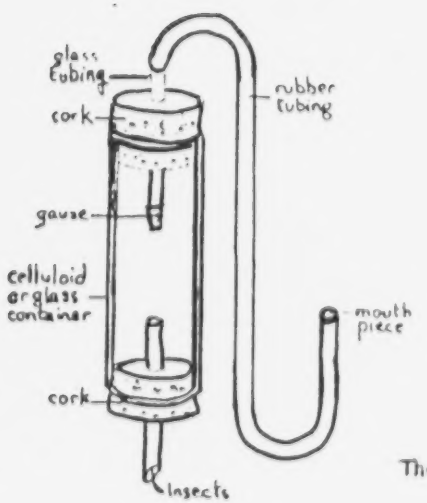


FIGURE 6

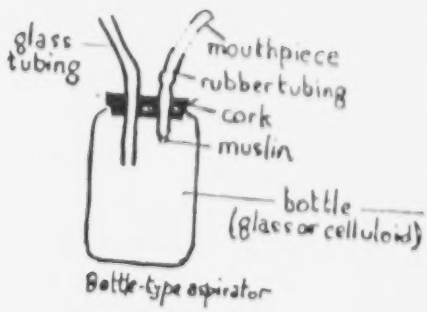
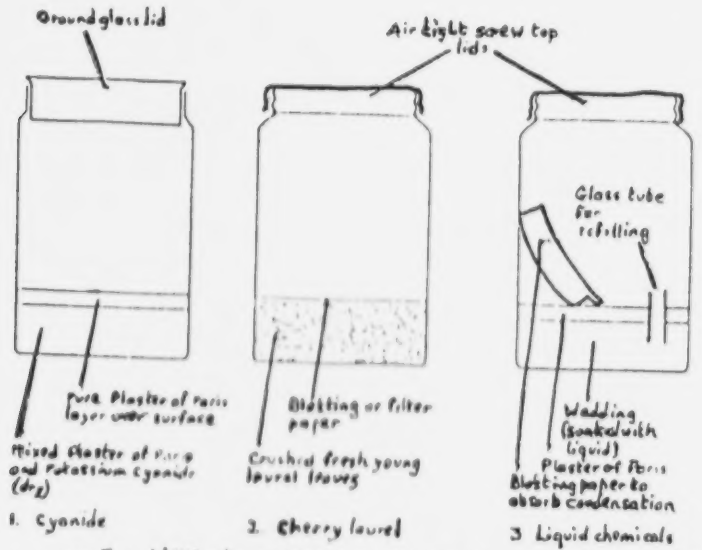
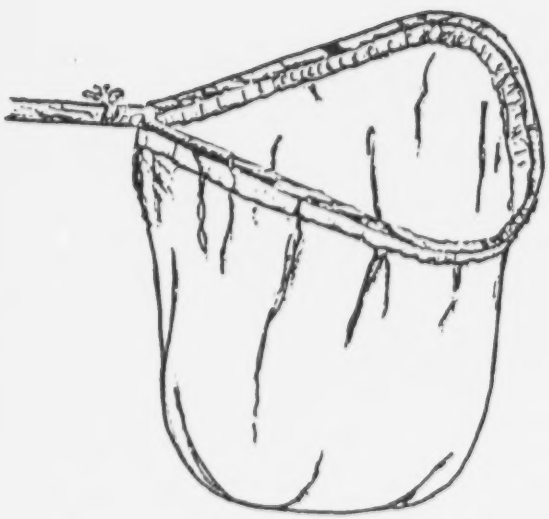


FIGURE 7

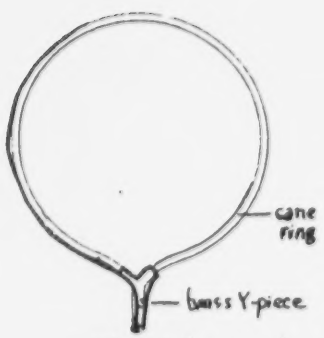

Ground glass lid
Air tight screw top lids
Glass tube for refilling
Pure Plaster of Paris layer over surface
Mixed Plaster of Paris and Potassium Cyanide (dry)
Blotting or filter paper
Crushed fresh young laurel leaves
Wadding (soaked with liquid)
Plaster of Paris
Blotting paper to absorb condensation
1. Cyanide
2. Cherry laurel
3 Liquid chemicals

Insect killing jars using (1) cyanide, (2) cherry laurel leaf, (3) ethyl acetate, or other similar liquid

FIGURE 8

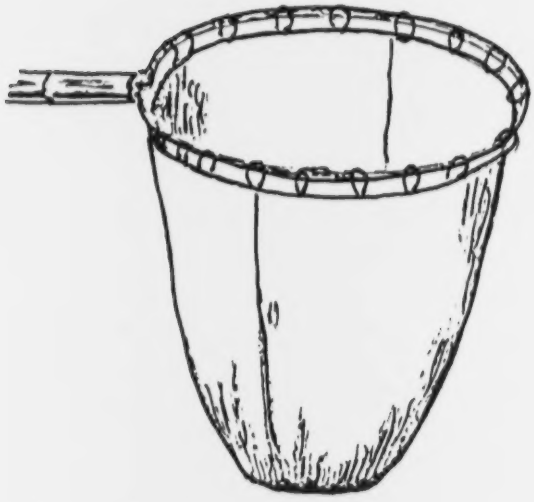


Kite net



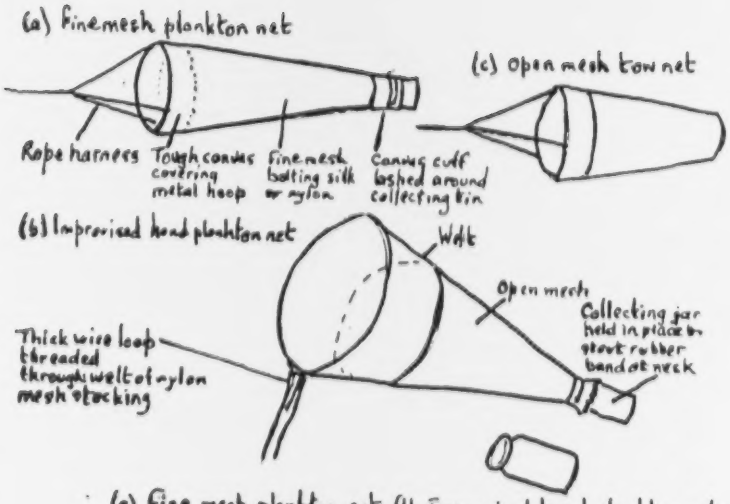
—Kite net frame made of cane, the net can be of any convenient width but is usually about 3 decimetres (1 ft)

FIGURE 9



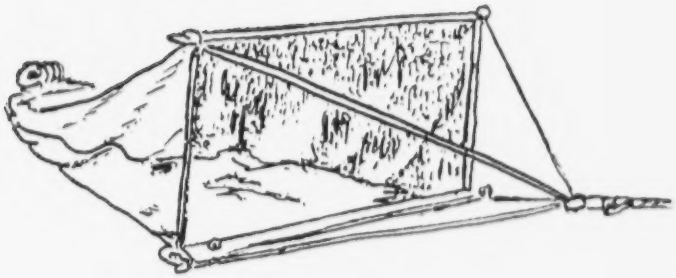
Sweep-net for use in low growing vegetation

FIGURE 10


(a) Fine mesh plankton net
(c) Open mesh tow net
Rope harness
Tough canvas covering metal hoop
Fine mesh bolting silk or nylon
Canvas cuff lashed around collecting tin
(b) Improvised hand plankton net
Wire
Open mesh
Collecting jar held in place by stout rubber band at neck
Thick wire loop threaded through welt of nylon mesh stocking

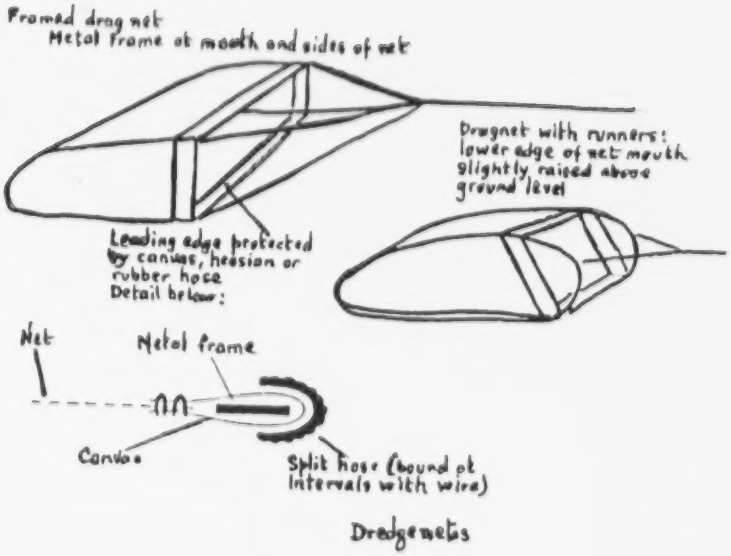
(a) Fine mesh plankton net (b) Improvised hand plankton net
(c) Open mesh tow net

FIGURE 11



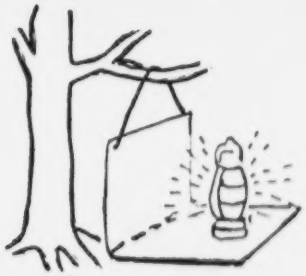
Drag net for use in water

FIGURE 12



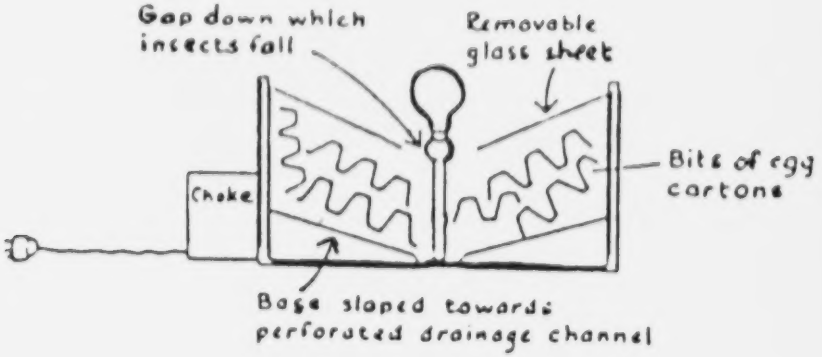
Dredgenets

FIGURE 13



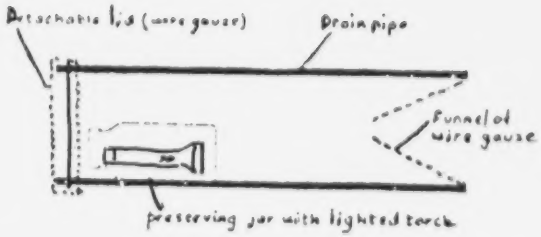
Simple light trap; illuminated sheet with both vertical and horizontal surfaces

FIGURE 14



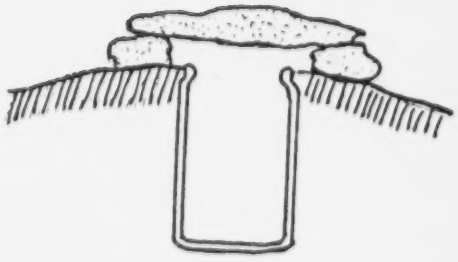
Mercury-vapour light trap

FIGURE 15



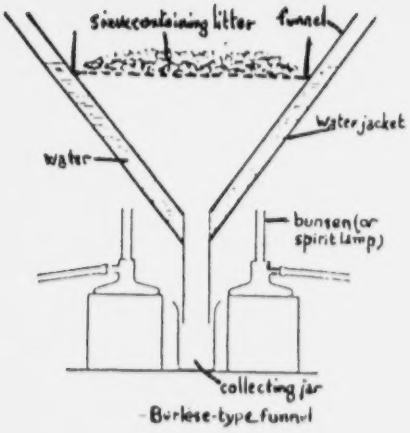
Drainpipe trap

FIGURE 16



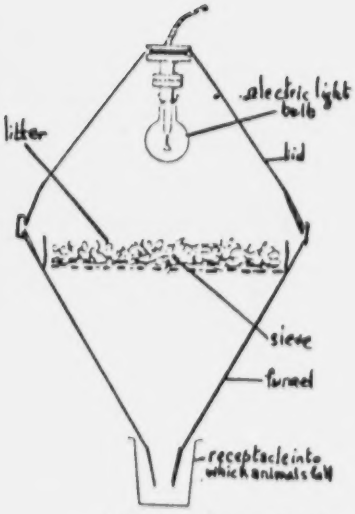
Pitfall trap

FIGURE 17



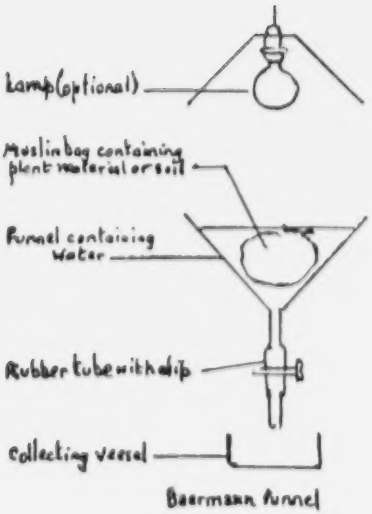
Burkese-type funnel

FIGURE 18



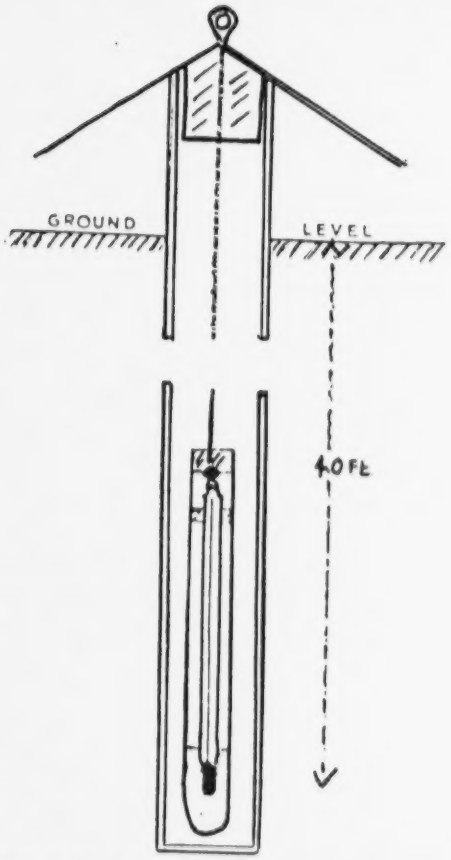
A Tullgren type of funnel

FIGURE 19



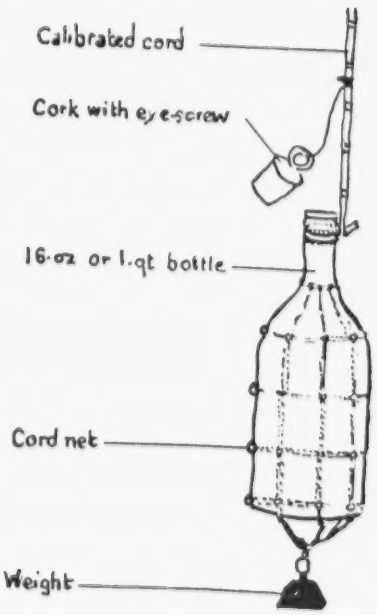
Baermann funnel

FIGURE 20



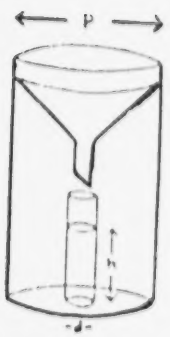
-Diagram showing the construction of the housing for a Soil Thermometer arranged to work 4 ft. below the ground.

FIGURE 21



Meyer Sampler Bottle

FIGURE 22

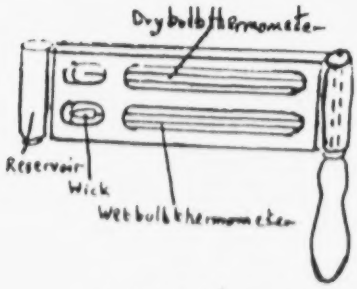


1 Rain gauge

FIGURE 23

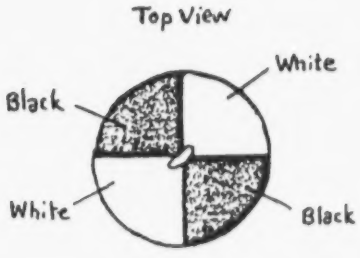
A typical Wet and Dry Bulb Thermometer. The wet bulb is kept moist by a wrapping of absorbent material which dips into a reservoir of water.

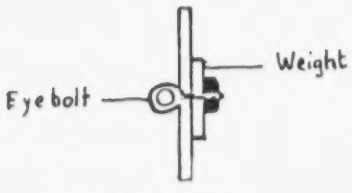
FIGURE 24



Whirling hygrometer

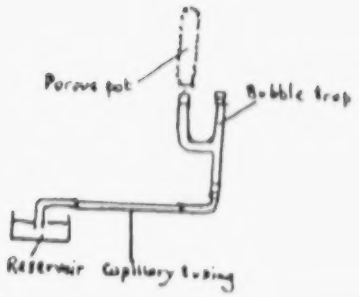
FIGURE 25





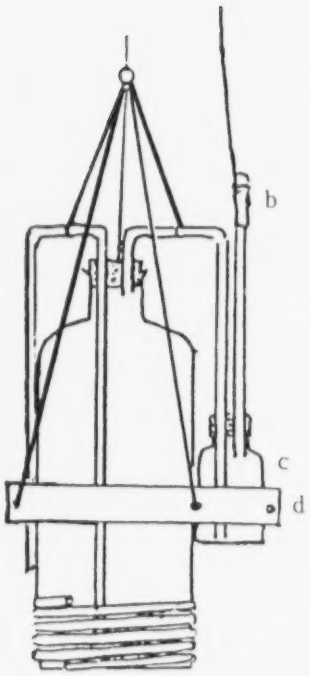
Secchi Disc

FIGURE 26



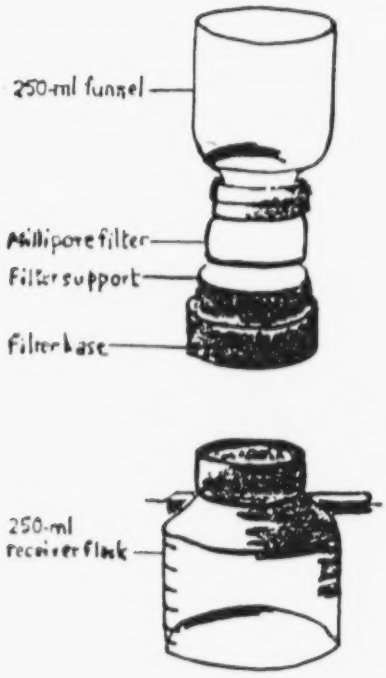
Darwin potometer in use as an atmometer

FIGURE 27



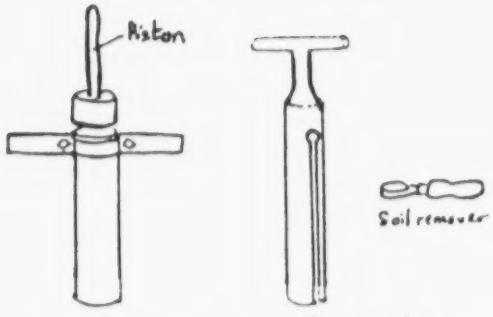
Sampler for use in deep water

FIGURE 28



Millipore Sterifil Filtration System

FIGURE 29



Apple-corer type samplers

FIGURE 30

CHAPTER 5

ETHOLOGICAL METHODS

Although it has developed as a separate scientific discipline from ecology, ethology (the study of animal behaviour) is an important aspect of ecological field studies. The distribution of animals and hence their relationships in an ecosystem is closely related to their behaviour. Animals are adapted to their environment to a large extent through their behaviour but also, through their behaviour, they can change the environment and hence alter the balance of ecosystems.

Studying animals in the field is difficult because, of course, they tend to move about. Thus the methods we use for studying plants or factors in the physical environment are not always appropriate. In particular it is not usually possible to go to animals and observe them; with many species they will just move away. The observer has to work out methods by which animals can be studied without disturbance but, at the same time, accurately and over appropriate periods of time.

Observations can be direct or remote. Direct observation can be undertaken from a hide, a camouflaged situation place near to where one expects the animals to be. Binoculars are valuable aids to direct observation as are cameras and tape recorders. Examples of remote observation are when one deduces the behaviour of animals by examining their effects on the environment e.g. eaten plants, footmarks and faecal or other deposits. Also apparatus can be devised to record behaviour indirectly. Thus a time-lapse camera set on a place where animals are expected to be will take photographs without the observer being there. An ordinary camera rigged so that it will take a photograph when triggered of by an animal hitting a string attached to it is another example.

Often a combination field studies and laboratory studies provides a suitable strategy for studying animal behaviour.

Field studies are necessary to provide information on (i) the stimuli each species normally responds to, (ii) the range of its natural behaviour (behaviour repertoire) and (iii) its overall behaviour organisation. Some activities are absent in captive animals but present in wild ones, e.g. aerial flights of some courting birds. In addition, laboratory conditions can produce abnormal behaviour; that is, behaviour that does not occur under natural conditions or occurs much less often e.g. self-mutilation.

The major advantage to laboratory studies is the elimination ('control') of the extremely large number of variables that affect behaviour. The need for controls in behaviour studies can be illustrated with the story of 'Clever Hans', a horse that could apparently count and spell. He used his right forefoot to tap out answers, even without his owner being present. It was finally discovered that the horse simply stopped tapping when the questioner unconsciously signalled (by relaxing suddenly or twitching) that the right answer had been reached.

A controlled experiment would have systematically varied aspects of the environment (such as the presence of the questioner) to eliminate them as possible influences.

There are two basic sets of controls, control by EXPERIMENTAL DESIGN and control of the ENVIRONMENT. The simplest control of the first type is a study where two groups of animals, randomly selected for membership, are treated identically except for one respect, the 'treatment'. Environmental controls include such things as (i) testing all animals at the same time of day, (ii) housing all in identical conditions, (iii) maintaining constant temperature, humidity, noise and lighting conditions during testing, and (iv) using standard handling methods.

In general, field work precedes laboratory work and is far more broadly based. Laboratory studies usually attempt to elucidate specific principles and do not attempt to describe broad patterns of behaviour. A good ethologist combines both approaches whenever possible.

Data Collection

Qualitative data

Description plays an important part in studying behaviour. Without an accurate description of what a piece of behaviour consists of determining how frequent it is or what causes it, for example, can be meaningless. For the ethologist a note book in which descriptions of behaviour are recorded and a bag of the procedures of study are kept is an essential tool. In keeping such records always record at once and write down as much detail as possible. What appears of little relevance at first may become very significant later.

The term used by ethologists for a description of the characteristic behaviour of a species is an ethogram. Preparing an ethogram or examining ethograms produced previously by researchers is the first stage in the study of behaviour. To provide an idea of what this entails we can use the words of a leading ethologist, Peter Marker.

> "The first task then is simply to observe the behaviour in general, in as many contexts as possible. Since we know that the surroundings can distort and modify behavior, the study should be made under conditions which are as natural as possible. Ideally we would follow the animal around on its daily routines in the wild state. In practice we usually have to compromise. A common solution is to cage the animal as comfortably as possible and allow it to behave in as natural a fashion as it will when left without disturbance. The bulk of behavioral data is gathered in this way, and it is supplemented by whatever field observations are possible. Gradually a picture is constructed of the motor patterns which the animal uses in its daily life, the stimuli---both physical and social---to which it seems to be responsive, and the ways in which

> the behavior changes with a shift in the physiological conditions of the animal. Even a partial picture is better than none at all as a pre-requisite for the selection of behavioural parameters for, say, a specific purpose.
>
> The collection of such data is the most arduous and demanding aspect of behavioral study. It requires that you spend much time watching the animals, exposing them to your own presence as little as possible. You will at first be confused by the great variety of complex movements, which follow so quickly upon each other that you cannot see what is happening. Then you will begin to see signs of order: the consistent use, by a bird or a mouse, of a certain posture for fighting or threat situations; a particular configuration of the shapes and colors of the fins of a male fish when it is courting a female. By recording data of this kind, however fragmentary at first, you will eventually begin to predict what an animal is going to do in a new situation, and to discern which behavior patterns are rigidly stereotyped, and which are variable. By relating this to variations in the circumstances under which the behavior occurs, you can begin to understand its motivation, and so on."

In order to make sense of animal's behaviour an ethogram must involve classification. The form of this will depend on the purpose of a particular study but it would be expected to include such behaviours as -

Feeding, drinking, breathing, defaecation and urination

Attack and defense; maintaining territory

Level of activity at different times in the day (and night?)

Mating and other reproductive behaviour

Social behaviour, including play.

Quantitative data

This is usally obtained by using coding sheets. With these activities are listed on a form, and the student simply checks each activity when it occurs. There may be a time element included on the form. The following is an example of a coding sheet used for recording behaviour of monkeys.

Time	Preaning	Eating	Sleeping	Fighting	Flying	Alert	
05							
10							
15							
20							
25							
30							
35							

Additional activities can be added to the sheet as they arise.

Some common measurements of behaviour that may be collected on coding sheets are:

Frequency per unit time.

Intensity of performance along some scale, e.g. intensity of bowing by pigeons.

Response after presentation of stimulus, e.g. movement of a mouse after being introduced to a strange environment.

Duration of performance e.g. duration of response to presence of predators.

Strength of stimulus required to elicit a performance, e.g. amount of food required to cause salivation.

Strength of stimulus required to inhibit a response or release an incompatible response.

Proportion of time one activity is associated with another.

Ranks of activities based on some criterion such as immediate survival value.

Frequency of interactions during a unit of time.

Frequency of proximate associations (within a certain distance).

Presence or absence of a behaviour.

Sampling

Methods of sampling the physical environment or static organisms such as plants (see page 20) are not always appropriate for studying mobile animal behaviour particularly when time has to be taken into account also. A discussion of different forms of sampling used by ethologists follows. With field studies at the school level it is not always possible to use refined sampling methods. However, a teacher when devising an appropriate simple method for their class should bear in mind the considerations research ethologists take into account.

There are four basic methods of time-sampling used by ethologists. First, sampling may be done in such a way that all possible samples have the same probability of being collected. This method requires the use of a table of random numbers, and is normally called RANDOM SAMPLING. It has little value (except in special cases) because of possible gaps in the records.

HAPHAZARD SAMPLING may or may not be less accurate - there is little grounds for assessing which. With this method, animals are observed at the researcher's convenience or when the animals are thought to be most active. It is a poor basis for descriptive studies because the observer may miss activities that occur at specific times of the day. It has no value for quantitative comparisons, except possibly for frequencies of interactions between certain groups of animals, for example in determining which individual is dominant on the basis of fights or threats that occur during a day.

REGULAR SAMPLING is sampling on a scheduled basis. It may or may not be BALANCED SAMPLING as well. Balanced samples are taken to cover all times of the day, weeks of the month, months of the year and to cover all the age and sex classes as well. For example, each week you may sample all the ages and both sexes at evenly spaced times over the whole day.

Sampling methods can also be classified on the basis of the type of data collected. In the AD LIBITUM method all observations are recorded whenever the observer sees the animals. These are usually also randomly timed and qualitative samples. They are the typical field notes. This type of sampling is only important when preparing a study or making preliminary observations. It has the disadvantages of possibly involving unconscious selection of easily observed activities as well as easily observed animals. It has, of course, no quantitative use, and is wasteful of time and effort. However, it can be valuable in beginning a broad-based behavioural-ecological study or with a new species.

The SOCIOMETRIC MATRIX (Table 2, page 63) is really a design form as well as a sampling method. In using it all interactions between pairs of animals are recorded. If only the frequencies of interactions between pairs are considered - for example, with birds when fighting, which one gave and which one received the most pecks - then this sort of data can be collected randomly, haphazardly or regularly. It is of course possible that the relationship changes with time of day or time of the year: therefore balanced sampling is best.

TABLE 2

SOCIOMETRIC MATRIX RECORDING FIGHTING BETWEEN BIRDS

		Winner		
		A	B	C
Loser	A	-	35	10
	B	23	-	50
	C	20	10	-

COMPLETE RECORD SAMPLING is an attempt to record the timing and duration of every occurrence of all categories of behaviour. Analyses may be of frequencies, durations, sequences or "budgets". Equipment which can be used include coding sheets, audio-tape recorders, and video-tape recorders. The limitations of this method are that observations must usually be short because of observer fatigue.

SEQUENCE SAMPLING consists of the observer recording the order of actions. If more than one animal is used, then the identity of the animals must also be recorded. A time element can be added.

TABLE 3

SEQUENCE SAMPLING

30 SECOND INTERVALS

	1	2	3	4	5	6	7	8	9
Animal No. 1	A	B	C	B	E	C	A	F	G
Animal No. 2	B	B	C	D	D	C	A	F	F

This type of sampling is usually used to assess the associations of activities of one individual or between individuals. The limitations are that many types of information are lost e.g. precise durations, presence of very short actions, and the observer may have difficulty observing more than one animal at a time.

FOCAL ANIMAL SAMPLING consists of observing one animal for a specified period, recording all its actions or a selected sample, then moving on to the next PRE-SELECTED animal. Generally the duration of the action, as well as its sequence and type, are recorded. The duration of each sampling period and amount of time the focal animal is out-of-sight must be recorded (for comparison with other samples). It uses the same procedures as complete record sampling and can be used in most behaviour studies but is particularly useful in the field where observations of more than one animal are extremely difficult.

INSTANTANEOUS SAMPLING is sampling behaviour at fixed intervals. The equipment used is usually simple: a watch, pencil and coding sheets. It has the advantage of allowing the observer to avoid making repeated decisions about when a behaviour changes from one action to another. It is not useful for studies of the duration of actions.

Some final things to bear in mind when studying behaviour. Firstly, a lack of enthusiasm will hurt the best designed study, but an abundance of it will almost invariably solve problems. Secondly, always try to get the best equipment and treat it with respect to get the best and longest use from it. Thirdly, always keep in mind ways of improving your design and record keeping but be wary about changing methods in the middle of a study. Finally remember observing animals requires patience.

Exploitation and Management (See Chapter 3)

a Exploitation of Natural Habitats
b Production
c Pollution
d Conservation

Type of Investigation

I Questioning and Observation
II Recording and Analysis
III Experimentation
IV Management Problem-Solving

Whilst the activities are arranged under the four environments it is possible to adapt those under one environment for use in others. The number of activities for the marine environment is particularly greater than that for the other three because several of the methods and some of the activities included can be adapted for use in the other environment.

CHAPTER 6

PRACTICAL ACTIVITIES

In order to provide direct, practical help to teachers a number of ecologists experienced in different environments were asked to construct a numberof fieldwork activities suitable for use with secondary school pupils. Each collection of activities is concerned with a particular environment, respectively marine, humid forest, savanna and desert.

Each activity is arranged in the following order.

Title

Objective - the knowledge and skills students should acquire and use during the activity.

Curriculum Links - topics usually tobe found in secondary school biology courses to which teachers can relate the activity and thus integrate it into their normal work.

Preparation - what the teacher has to do before starting the activity to ensure its success.

- the knowledge, skills and attitudes students require to undertake the activity.

- equipment and other materials required.

Strategy - the procedures to be undertaken by the teacher and students during the activity.

Follow-up - ideas for extending the activity and linking the activity to further studies.

Classification of Activities

Following the title of each activity is a collection of letters and numbers in brackets. These refer to the major ecological principle(s), aspect(s) of exploitation and management and type(s) of investigation covered by the activity. They are classified as follows.

Ecological Principles (See Chapter 2)

A Diversity and Distribution
B Interrelationships
C Change and Equilibrium
D Movement and Behaviour

THE MARINE ENVIRONMENT

INTRODUCTION

Many schools in the tropics are close to coastal marine environments. The oceans contain many forms of life that are of great interest in biology. Life first evolved in the sea, and many primitive types of plants and animals occur there. Any school within reach of the sea should take advantage of this great natural laboratory.

The intertidal zone along the shore that is alternately covered and uncovered by the tides is the easiest area to reach for class field activities. Almost every activity in this section can be done by pupils on the shore when the tide is out. A few can be extended into shallow water by students who are good swimmers. In areas with little tidal variation, it may be necessary to wade into shallow water to observe features of interest.

There are four major habitats in tropical intertidal areas: sandy-muddy areas, mangroves, coral reefs and rocky shores. Only one or two of these will be available to most schools, but enough activities are proposed to demonstrate basic biological concepts in any of these environments.

Sandy and muddy habitats have a substrate of fine sediment that is too mobile for many organisms to attach to, particularly in the wave zone. Seagrasses or seaweeds may root in the sand or attach to small rocks and shells, and many animals will live on or burrow in the sediment. It is often necessary to dig up the top layer of sand or mud to discover this interesting community.

Mangrove trees are specially adapted to growing in salty water rooted in anoxic sediment. They are to be found mainly in estuaries and muddy coastlines where freshwater and saltwater mix, and each species may have precise salinity requirements. Their roots help to trap sediment and create a protected environment, while providing a place for many algae and animals to attach and live. Mangrove leaves are the beginning of an important detrital food chain.

Coral reefs are major geological structures built by biological activity through the accumulation of the skeletons of corals and coralline algae and of sand made from algae and animal shells. The coral reef ecosystem is very complex, with more different species crowded into a small area than almost any other habitat on earth.

Rocky shores are less common in the tropics than in temperate areas. Their hard substrates provide attachment for many kinds of plants and animlas, which compete for space in the intertidal and high subtidal. Many man-made shorelines consist of concrete walls, large rocks or pilings which may develop similar communities to rocky shores when they have been in the sea long enough.

Life is not easy in the intertidal zone. While light is abundant and nutrients are often available, it is necessary to attach to something to keep from being swept away by the force of waves and currents. Space is often at a premium, leading to fierce competition. It is necessary to withstand extreme temperatures and alternate wetting and

drying, as well as salinities from fresh rainwater to pure salt crystals. Growth can be rapid, but predators are numerous. The organisms of the intertidal show many different strategies for adapting to this extreme environment.

Class field work in the intertidal also presents some special problems and dangers. Where there is a small tidal range, it may be necessary to wade and to look at the bottom through the water's surface. Reflections and water movement can make this difficult, and pupils may need to use glass-bottomed buckets or boxes, facemasks or goggles to see clearly. Where the tidal range is great, the intertidal will be more accessible at low tide, but attention must be paid to the incoming tide which can rise quite rapidly. Obtain accurate tide data so that field work can be planned for a low tide period. Start either at low tide at the water's edge and work towards the shore, or follow the low tide out and in. Waves can also surprise unwary students, so care must be taken to choose conditions and sites with an adequate margin of safety. Students must always be supervised closely while in the field.

Some dangerous animals may be encountered in coastal waters as on land, so teachers should inform themselves about those which occur locally (cone shells, stone fish, etc.) and warn the pupils about them before going into the field. Many animals have sharp shells or edges, and care must be taken to avoid cuts and scrapes. Pupils must wear adequate footware such as shoes of rubber or canvas with soles of sufficient thickness to protect feet from cuts, spines and stings. Protection from the sun may also be advisable. A first aid kit should always be at hand just in case. With these simple precautions, a class field trip to the intertidal can be a safe and rewarding experience.

1. COLLECTIONS OF MARINE LIFE

(A; I, II)

Objectives

1. to observe marine plants and animals closely.
2. to prepare herbarium mounts and other speciments.
3. to identify organisms.
4. to understand the principles of classification.

Curriculum links

Identification and classification, species, biological diversity.

Preparation

Habitat: any accessible coastline, preferably with a hard bottom and attached plants and animals.

Time required: minimum 2 hours in the field, not counting the time to reach the coastal habitat, plus 2 - 3 hours in the classroom or laboratory to work on the collections.

Teacher: obtain and study whatever reference works and keys are available locally for the identification of marine life. Illustrated works are better for student use. Decide what should be collected and what the collections will be used for.

Students: the major groups of marine plants and animals should be studied before the field activity.

Equipment:
small knives for prying organisms off hard surfaces
shovels or trowels for digging in sediments
small plastic bags for collecting specimens
small bottles or vials for fragile specimens
buckets for carrying specimens and equipment
small pieces of heavy paper for labels
cards or heavy drawing paper for herbarium mounts
pencils
hammer and chisel (supervised by teacher)
small dip nets (made from wire and mosquito screen)
shallow trays or dishes for sorting
old newspapers
pieces of muslin to cover herbarium mounts
aquaria or large glass jars
hand lense
microscope or dissecting microscope (optional).

Strategy

Question: What plants and animals live in the coastal marine environment?

1. Collecting organisms is an excellent way to learn to observe them closely. However, the amount of collecting by the class must be carefully controlled by the teacher, as a large class collecting too many organisms can have a serious effect on the environment. Organisms should not be collected except for a definite purpose.

2. Each pupil or small group of pupils is supplied with collecting tools (knife, shovel, dip net), bucket, plastic bags, bottles, labels or notebook, pencil.

3. Pupils should be instructed to make a collection of certain common organisms such as:

   algae and seagrasses
   small common molluscs or shells
   small crabs, crustacea, isopods.

4. In addition, one example of each plant and animal observed can be collected for the whole class. Duplicate specimens should be returned to where they were collected.

5. For each collection, a label should be made with the following information: name of collector, date, place name, where the organism was found, type of substrate or habitat, associated organisms, size of the whole organism (if only a part was taken), relation to tide level. The organism and label should be put together in a plastic bag or bottle. Alternatively, the specimens can be numbered and the information written down in a small field notebook. Space should be left under each entry to add the name and other data once identification is made.

6. Algae and most animals should be kept damp but not under water. Only swimming animals should be kept in water. Avoid overheating in the sun.

7. If possible, the class should return immediately to the school laboratory or other work area to sort and prepare the collections and to observe the living animals. If more than 1 day will pass before the collections can be worked on, they should be held indoors in large containers or aquaria of seawater or preserved in 3% formaldehyde in sea water. Preserved collections should be rinsed before being given to the students for sorting. Formaldehyde should not be used in containers or work areas which will serve for holding or rearing live organisms, as the fumes and residue are toxic to delicate organisms.

8. In the laboratory, collection should be sorted and identified, with the names and other information added to the labels or notebooks. If references giving the scientific names are not available, then common names or local names should be used.

9. If time permits, observe the special features, morphology and behaviour of the organisms collected. Place the organisms in trays or aquaria to make observation easier. Examination with a hand lense or microscope can also help students to see smaller organisms and features.

10. Specimens can be prepared for a permanent class collection. Algae are laid out or floated out on heavy paper or cards, then covered by a piece of muslin and dried between several sheets of newspaper in a plant press or under a heavy weight. The newspaper should be changed every day until the algae are dry. Interleaving with corrugated cardboard can speed drying. Shells are cleaned, and most other hard-bodied animals can be air-dried (outside, they will smell!). Soft-bodied animlas must be preserved in 70% alcohol.

11. Pupils can also make personal collections of the common algae, shells and crustacea they have collected.

12. The completed collections should be arranged according to the scientific classification of organisms, if that is available. The teacher should explain the distinguishing characteristics of the organisms. If a scientific classification is not possible, an artificial classification can be made as follows:

    - Decide on the criteria for separating the organisms (colour, shape, anatomy, etc.)

    - Separate and classify the organisms according to the criteria adopted.

    - Make any revisions in the criteria that seem necessary.

13. On the basis of the complete collection of all the plants and animals observed, make a species list for the area showing how many different kinds of organisms occur there.

Teaching hints

There should be no uncontrolled class collecting, as this can rapidly strip an environment. Too often animals collected by classes are not looked at again but are left to rot in various containers in the laboratory. This should be prevented. There should be a definite purpose in mind for any collection made. Restrict multiple student collections as much as possible to organisms that are very abundant and are known to regenerate quickly.

It is not necessary that the identifications and classifications made by the class always be scientifically accurate as this often requires reference to complex scientific works that are inappropriate at the second school level. It is the scientific methods of observation, identification and classification that should be understood. However, students should be encouraged to differentiate between forms at the species level.

Follow up

This activity is a good preparation for many of the ecological activities that follow, as it helps to ensure that different types of organisms are familiar to the students.

2. SANDY-MUDDY SHORE ZONATION

(A, B; I, II)

Objectives

1. to discover the organisms that live on or in sand and mud.
2. to observe the relationship between environmental factors or gradients and organism distributions.
3. to learn the principles of quantitative sampling.
4. to process and present numerical data and to test it for statistical significance.

Curriculum links

Identification of organisms, distributions, gradients of environmental factors, sampling and significance.

Preparation

Habitat:	a beach, mud flat or other coastline of sand, mud or other fine loose material, preferably exposed at low tide.
Time required:	1 - 3 hours in the field plus 2 - 3 hours processing the samples in the laboratory or classroom.
Teacher:	obtain references to local marine life. Select simple statistical tests for analysing the data.
Students:	introduction to the kinds of marine life to be found, to types of organism distributions (regular, random, clumped) and to principles of sampling and statistical analysis.
Equipment:	rope or string to mark transect quadrat sampling frame (square 25 cm x 25 cm or larger, or circle of plastic gardenhose up to 1 m diameter) rulers 20 or 30 cm plastic bags for samples buckets strong shovels or trowels sieves of different mesh sizes (e.g. 2 mm, 5 mm, 10 mm) pans or jars for sorting and observing organisms.

Strategy

This activity can be undertaken qualitatively (Question 1) and/or quantitatively (Question 2), depending on the level of the class and the time available.

Question: What is the zonation of the plants and animals on or in the sand or mud?

1. Lay out a transect across the intertidal from land to sea with 10 marks at regular intervals from the high tide line to the water's edge. Use a string or rope with evenly spaced marks, or use a tape measure and work across the area in segments.

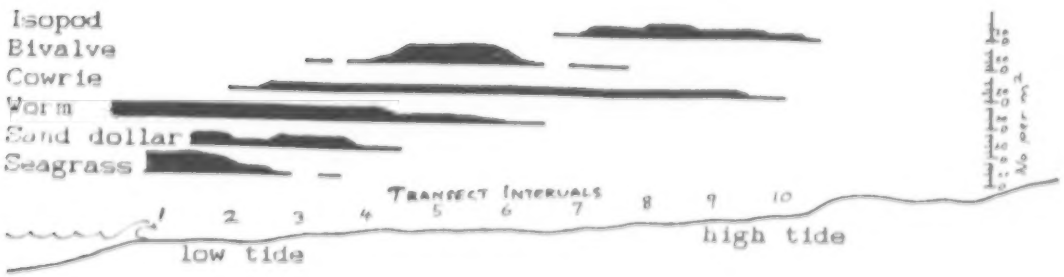
2. Starting at the seaward end at low tide, mark a quadrat sampling area at each mark using the square or circle frame, and note the kinds and numbers of all the organisms visible at the surface. Within the quadrat, or in a subsample of 25 by 25 cm if the quadrat is bigger, dig up the sand or mud to a depth of 10 to 20 cm together with any plants or animals in it. The sample can be washed through sieves immediately (see 3 below) or taken back to the school for washing and sorting. Keep each sample separate in a plastic bag, pan or bucket, and number the samples in order.

3. As you go along, or else later in the school laboratory, wash each sample through at least 2 sieves of different mesh sizes (say 10 mm and 2.5 mm). Note the kinds and relative abundance of organisms in each sample.

4. Prepare a chart showing where each organism occurs along the transect. In the example shown in Figure 31, the thickness of the line shows the abundance of each organism. Try to relate the distributions observed to known physical factors or gradients.

FIGURE 31

TRANSECT WITH ORGANISM ZONATION



Question 2: What is the distribution and abundance of organisms across the intertidal zone?

5. Lay a transect marked at 10 regular intervals from high water mark to the water's edge at as low a tide as possible, and take 5 samples at each mark at random distances from the line. Each sample should consist of the sediment and organisms in a 25 cm x 25 cm quadrat removed to a depth of 10 - 20 cm, and should be kept separate and numbered according to its location.

6. The samples should be sieved and sorted by kind of organism, and the numbers of each organism recorded for each sample. Prepare a table showing the distribution of organisms across the transect.

7. The significance of the differences observed between samples at the same mark, and between marks, can be tested statistically, either with manual methods or by processing the data on a microcomputer. Try to explain the differences in distribution and abundance between organisms.

Teaching hints

The size of the quadrats sampled will depend on the organisms present. For large organisms such as sand dollars, starfish and sea cucumbers, it may be necessary to mark off a 2 m by 2 m square, or even a 4 m radius circle defined by turning a rope around a stake. For small organisms (worms, small clams, etc.) the 25 cm quadrat may be sufficient, or an intermediate size (50 cm by 50 cm, 1 m square, or 1 m diameter circle) may be needed, depending on experience in the local environment.

On a coral reef lat, the underside of loose coral blocks can be a good sampling space. Measure the block and count all the attached organisms on the top and bottom. Standardise the counts per unit of area later.

A microcomputer equipped with data-base management (or spread-sheet) software can be used to construct tables and perform graphic analyses. Statistical tests like analysis of variance and regression analysis can also be done easily with standard commercial software.

Follow up

For an experiment to test some of the factors controlling animal zonation, see Activity 8, Translocating Molluscs.

3. THE MANGROVE COMMUNITY*

(A, B, C; a; I, II, IV)

Objectives

1. to observe and record the diversity and distribution of populations associated with gradients in physical factors.
2. to study interrelationships among organisms and between organisms and the physical environment.
3. to understand spatial relationships in the mangrove ecosystem.
4. to consider the effects of disturbing the mangrove ecosystem.
5. to understand the need for conservation of mangrove areas.

Curriculum links

Community, habitats, biological factors, physical factors.

Preparation

Habitat: a typical coastal mangrove area; if possible both undisturbed and disturbed mangrove areas can be studied and compared.

Time required: 2 - 3 hours of field work and a similar time for laboratory study of collections and data analysis.

Teacher: select appropriate sites for the surveys where the slope of the land can be measured in a straight line. Divide students into small groups (up to 4 - 5), with one group to measure the slope of the shore while the others make records at each station.

Students: a general introduction to mangrove forests would be useful. Special problems of working in mangroves, such as not walking in others tracks (because of the danger of sinking deeper in the mud) and keeping close to a supporting plant, should be understood.

Equipment:
rope or string
stakes
plastic buckets (1 per group)
plastic bags (5, 10, and 20 litres) and rubber bands/ties
marking pen
thermometer
pH paper
paper labels (3 x 5 cm)
sieve or screen (1 mm gauge)
jars (250 cc)

---

*Based on an activity devised by Dr. Pisam Soydhurum.

scissors
forceps
small shovel
quadrat sampling frame (25 cm square or larger)
field notebooks and pencils
level or long clear plastic tube filled with water
measuring tape
ethyl alcohol (optional)
plant press (optional)

Strategy

Question: How are mangrove ecosystems and the organisms that live within them different from other ecosystems?

1. Lay a straight transect line with rope or string from the water's edge to dry land above high tide level. Place stakes at 10 metre intervals along the line to mark a station for each group of students. Number the stations.

2. To sample the animals at each station, throw the quadrat at random in the vicinity of the stake. Observe and record the kinds and numbers of animals found on the surface or underground within each quadrat. Put examples of each animal in one plastic bag, and a soil sample in another, and label them with the station number. Also record the kinds and numbers of animals or plants (especially mangrove roots) nearby. Throw the quadrat again and make another count and sample of speciments.

3. For the plants, mark off squares 10 x 10 metres along one side of the transect. Within the square, count and record the names and heights of all plants with a diameter of 4 cm or more. Collect samples of each plant, including leaves, flowers and fruits if possible. Record the animals found on each part of the plant.

4. Record the physical factors at the station:

   - characteristics of the soil (colour, odour, hard or soft)
   - light intensity (estimated qualitatively)
   - pH of the soil using pH paper
   - air temperature
   - soil temperature at 5 - 10 cm depth (allow 2 minutes)
   - slope or inclination (using tape measure and sight level or water level in transparent plastic tube).

5. After the field work or in the laboratory, wash the animals by running water over them in a sieve or screen. If a voucher collection is needed, preserve the animals in jars of alcohol; dry plant specimens in a plant press. Examine the plants and animals (with the help of a hand lense or microscope, if available), and identify them if possible.

6. Prepare a table listing for each station the name, density (number per square metre), height (for plants) and observable characteristics of each animal and plant on the surface or underground.

7. Make a table for the physical factors, showing for each station the soil characteristics (colour, odour, soft or hard), soil temperature, air temperature, pH and light intensity.

8. Draw a diagram showing the slope of the area studied in cross-section, and enter the information from the table of plants and animals to show their distributions along the transect in order to demonstrate the spatial distributions in the ecosystem. (Figure 32, page 78)

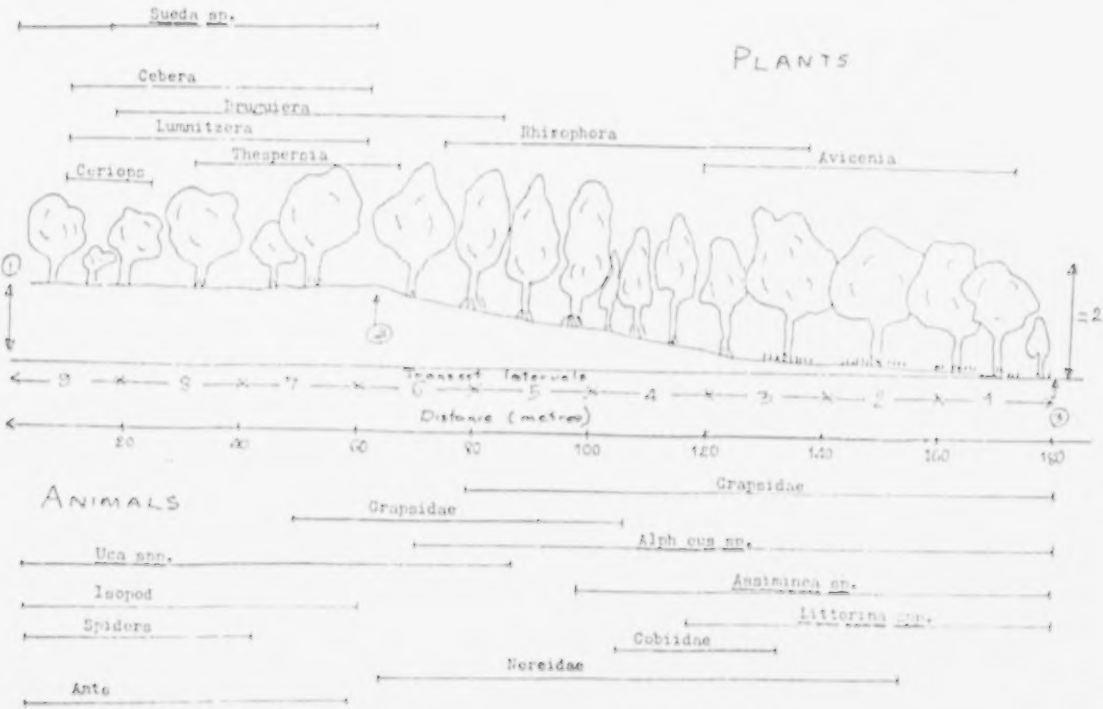
9. If a disturbed mangrove site is available, do the same survey in the disturbed site and compare the two transects.

10. Discuss with the class what may happen with different kinds of mangrove disturbance (logging, filling, polluting, changing water circulation).

11. Review with the class the values of mangroves and the importance of their conservation.

FIGURE 32

PLANT AND ANIMAL DISTRIBUTIONS IN A MANGROVE ECOSYSTEM



Follow up

The total picture of the mangrove ecosystem formulated from the data in the field activity could be compared with data from other classes or from other areas. Any similarities or differences should be discussed.

4. CORAL REEF STRUCTURE

(A, B; I, II)

Objectives

1. to observe the structure of the coral reef.
2. to relate the reef structure to some environmental factors.
3. to study the role of reef-building organisms.

Curriculum links

Community structure, organism-environment relations (biological and physical factors), skeletal accumulation.

Preparation

Habitat:	an accessible coral reef where pupils can observe as many different reef zones as possible in safety; to be surveyed at a very low tide.
Time required:	2 hours in the field.
Teacher:	obtain information on the tidal range, water circulation and storm effects on the reef to be studied; know the tide level at the time of the survey. An aerial photograph of the reef would be useful if available.
Students:	a general introduction to reef types (fringing, barrier, atoll) and reef zones (back reef, reef flat, reef slope, etc.) and the types of reef organisms.
Equipment:	long tape measure (30 m +) or rope marked every 2m pole 2 m long marked at 10 cm intervals thermometer carpenter's bubble level (attached to a board if short) field notebooks and pencils.

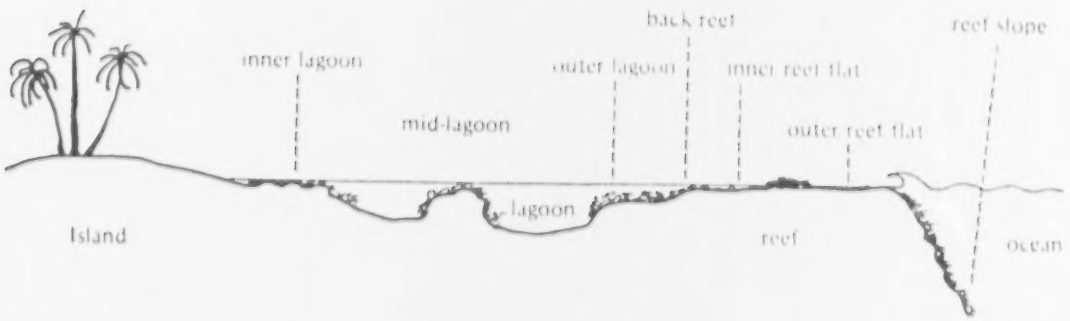
Strategy

Question: What is the structure or form of the coral reef?

1. The best way to visualise the structure of a coral reef is to draw a vertical profile (Figure 33). Select a typical area of coral reef, and establish a starting point on the shore. Lay out the tape measure or marked rope seaward from the starting point, and follow this transect line out recording the type of bottom and organisms and their distance from the starting point. It may be necessary to move the tape or rope several times to cross the whole reef.

FIGURE 33

CORAL REEF PROFILE WITH ZONES



2. To determine the vertical dimension (depths) along the profile, position the carpenter's level on the shore above the highest point on the reef so that it is perfectly level and pointed out along the transect. Using the calibrated pole held vertically on the reef, it should be possible by line of sight back to the level to determine the approximate depth of the reef features recorded along the profile.

3. Note the important reef-building organisms along the profile (corals, coralline algae, etc.). Have the students observe in which zones the most reef construction is taking place. Try to recognise the pieces of organisms that make up the sand and rubble.

4. Take the temperature of the water in the lagoon, beyond the reef, and in any pools of water trapped at low tide. Note which areas are exposed to air, sun and rain at low tide, which areas receive the force of breaking waves, and where water circulation is the best for animals feeding on plankton in the water.

5. Have the students imagine what happens on the reef during a major storm. Is there evidence of storm damage, such as large rocks, rubble banks or other accumulations on or behind the reef?

6. From the measurements and observations made, the class should prepare a profile of the coral reef, labelling the zones observed and relating them to physical factors. As many pertinent details as possible can be added to the profile.

7. If an aerial photograph of the reef is available, relate the profile of the reef to its overall shape and note the distribution of reef zones.

Teaching hints

The preparation of the profile can be an individual or group exercise. If the class has access to different types of reef, profiles of these different types can be made and compared.

5. ROCKY SHORE ZONATION

(A, B; I, II)

Objectives

1. to discover the organisms that live on a rocky shore.
2. to observe the relationship between environmental factors or gradients and organism distributions.
3. to look for examples of competition for space.

Curriculum links

Identification of organisms, distributions, gradients of environmental factors, zonation, competition.

Preparation

Habitat:	a rockyshore, seawall, wharf piling or other hard substrate across the intertidal zone and accessible at low tide.
Time required:	up to 2 - 3 hours, depending on the width of the intertidal.
Teacher:	obtain references to local marine life and tidal cycles.

Students: introduction to the organisms of rocky shores; explanation of tides and their effects on exposure in the intertidal.

Equipment: tape measure
field notebooks and pencils

Strategy

Question: What is the distribution of marine life in the intertidal zone?

1. Many organisms try to crowd onto a rocky shore where they can attach and receive light, water and nutrients. They usually occur in horizontal bands or zones in the intertidal zone depending on how often and how long they are exposed to air. The class should follow a low tide out and in, observing and making notes on the zones of organisms observed.

2. If the shore is steep and nottoo irregular, measure the width of the zones and their height relative to the tide level with a tape measure.

3. Note the common organisms that are the best indicators of each zone. Do some organisms have wider tide ranges than others?

4. Prepare a diagram of the zonation observed relative to tide levels.

5. On the basis of the observed zonation of organisms and the information on tidal cycles, the class should determine the length and frequency of exposure to the air for each zone identified.

Follow up

The activities on functional morphologies can be used following this activity to explain some of the adaptations that allow organisms to survive the conditions in each zone.

6. ZONATION ON MANGROVE TRUNKS

(A, B, C; I, II)

Objectives

1. to discover the organisms that live on mangrove trunks and prop roots.
2. to observe the relationship between environmental factors or gradients and organism distributions.
3. to appreciate the importance of a place to attach in an environment where attachment is difficult.
4. to observe how organisms can create an environment for other organisms.

Curriculum links

Identification of organisms, distributions, gradients of environmental factors, zonation, interrelationships.

Preparation

Habitat: mangroves with accessible trunks or prop roots well colonised by benthic organisms.

Time required: 1 - 2 hours of field observations.

Teacher: select a good low tide when trunks will be accessible; obtain references to local marine life.

Students: an introduction to mangrove ecology; an explanation of tides and their effects on exposure in the intertidal.

Equipment:
Knife or scraper for collecting
bucket, plastic bags or small jars
metre stick or 2m tape measure
field noteboosk and pencils
hand lens or microscope

Strategy

Question: What organisms live on mangrove trunks and how are they distributed relative to the tide level?

1. The soft, muddy bottoms of mangrove coastlines are not good places for many plants and animals to attach. Mangrove trunks and prop roots are often the only place where algae can stay in the light and animals keep out of the smothering mud. Such places are thus often crowded with life. However, the rise and fall of the tides covers and uncovers different levels of the trunks for different lengths of time, producing a vertical zonation of organisms.

2. Measure the width of each zone or band of organism distribution relative to the tide level.

3. Collect samples of each organism or association for identification on the shore or in the laboratory. Examine the algae and small animals with a hand lens or microscope. Determine the names of as many organisms as possible.

4. Combine information on the levels of each zone and on the tidal cycle to calculate the frequency and duration of exposure for each zone.

5. Prepare a diagram showing the zonation of common organisms on a mangrove trunk relative to the tide levels.

6. Try to explain the distribution of organisms in terms of their resistance to exposure to air, their vulnerability to predation by animals in the water, or other factors.

7. If there are few or no organisms at certain levels, try to explain why (changing heights of the sediment, grazing, pollution, storm scour, sedimentation, turbidity, etc.).

8. If time permits, experiment with the effects of changes in level on organism distributions. Select three similar prop roots or small trunks with the same zonation patterns. Cut off two of them above and below the main intertidal zones, and reattach them to one side of the third (the control), displacing one 20 - 30 cm (or more if the zones are wide) above its normal level, and the other the same distance below its normal level. The roots can be wired or nailed into place.

   Return to examine the changes in organism distributions and zones on the two displaced roots relative to the control, after 1 day, 3 days, 1 week, etc. until the normal zonation is re-established. Measure and note the changes observed on each visit. How long does it take each organism to readjust to its normal zone? Do some move while others disappear and grow back again? Do some organisms persist outside of their usual zones? How do you explain these observations?

7. AREA COVERAGE ON A CORAL REEF

(A, C; c, d: II)

Objectives

1. to observe the area of a coral reef covered by different types of plants and animals.
2. to learn the importance of semi-quantitative observations.
3. to estimate the relative importance of different groups in productivity and reef construction.

Curriculum links

Community structure, productivity, competition for space, environmental impact assessment.

Preparation

Habitat: choose a shallow coral reef area, preferably not steeply sloping, with good coral cover for the area, such as a reef flat or shallow back-reef.

Time required: 3 hours.

Teacher: the categories of bottom and organisms to be quantified may need to be adapted or simplified for local conditions.

Students: the categories to be measured and the method for estimating percent cover should be studied carefully before the field activity.

Equipment:

steel stakes or rods 1 m long
heavy hammer
or
ropes or cords with loops at each end, 4 m long
tape measures 30 - 50 m long
field notebooks or underwater slates and pencils
face masks or goggles (snorkles and fins desirable)

Strategy

Question: How important are the different plant and animal groups on a coral reef?

1. Divide the class into groups of 2 - 5 students. One or more students shoudl make observation while another records the results. Measures by different students can be recorded separately and cross-checked for consistency. Select one survey site for each group, spaced some distance apart. Two alternate methods can be used to estimate percent cover, as follows:

2a. **Estimation within a circle.** Drive a steel stake into the centre of each survey site. Put one loop of the 4 m rope over the stake. The circle described by the rope as it pivots around the stake defines a survey area of 50 square metres. Observing the bottom as the rope passes over it helps to make observation of the area more systematic. The estimation of the percent cover, or area of the circle occupied by either a class of sediment or a group of organisms, requires a certain educated judgement. Six classes coded 0 to 5 can be used:

0 = none (0%)
1 = a little (1 - 5%)
2 = some (6 - 30%)
3 = nearly half (31 - 50%)
4 = more than half (51 - 75%)
5 = almost all (76 - 100%)

It is difficult to make accurate judgements for the area covered by many small scattered things, a few big things, and things that are unevenly distributed. Figure 34, page 87, illustrates some different amounts and types of area coverage.

2b. **Line intercept method.** A tape measure 30 to 50 metres long is laid out parallel to the reef or shoreline. The group goes along the line recording the number of centimetres covered by each type of bottom, and then a second time recording the types of organisms. The total number of centimetres for each group, divided by the length of the line, gives the percent coverage for that group. If possible there should be enough tape measures for each group to have their own line, or several lines can be done sequentially and compared.

3. In looking at the bottom, there will be sand or bare rock in some places, and corals, plants or other things that are attached or fixed in place on the bottom. Two important measures of area coverage will be made: bottom type and major biological groups.

4. The first measure is what the bottom is made of. Much of a reef is solid, hard rock all cemented together, but in some places, and particularly on the lagoon floor, there is loose rock or sand on top of this. Measure what is seen at the surface, not what may be underneath if you dig down. The loose material or sediment is subdivided into 4 types based on the size of the individual grains, giving 5 categories of bottom type:

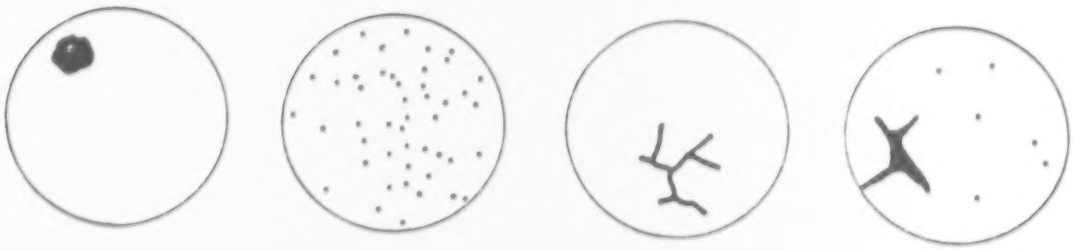
mud: very fine and soft, individual particles not distinguishable;

sand: small bits of rock or shell with the separate grains visible, but smaller than rubble;

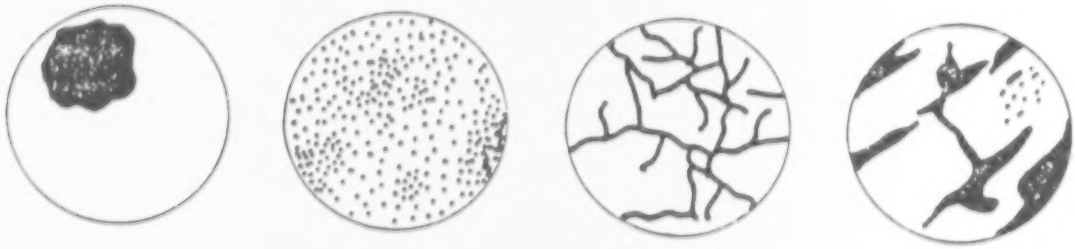
FIGURE 34

CLASSES OF PERCENT COVER

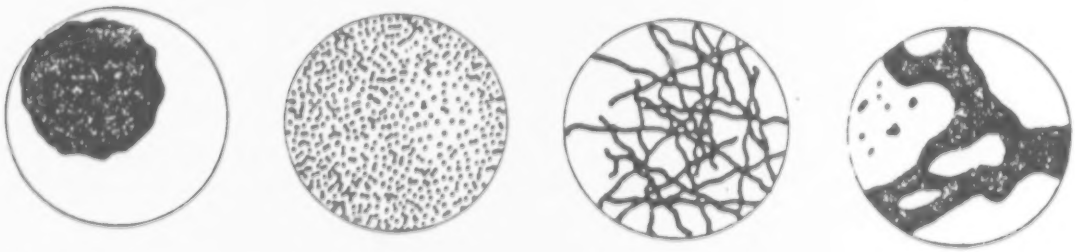
1 = 1 – 5% : a little



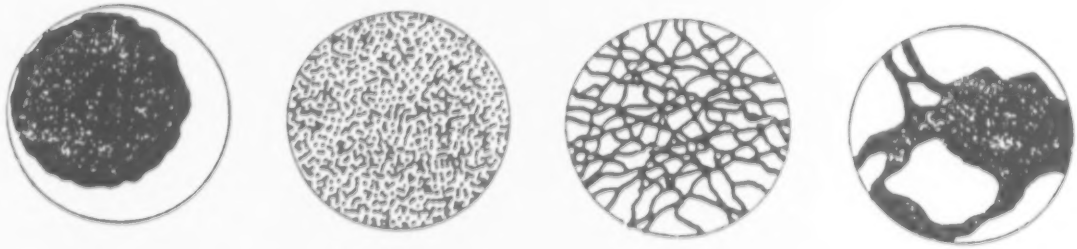
2 = 6 – 30% : some



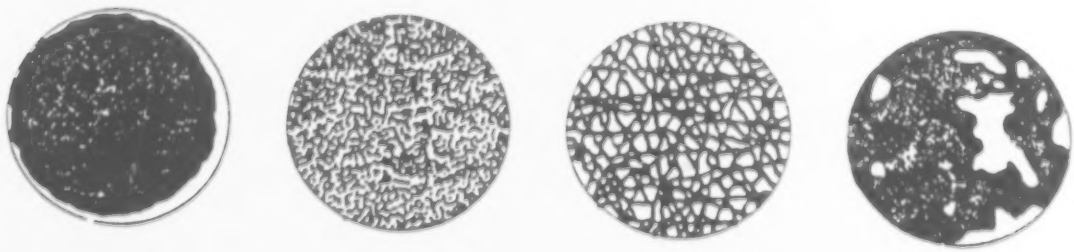
3 = 31 – 50% : nearly half



4 = 51 – 75% : more than half



5 = 76 – 100% : almost all



rubble: pieces of broken coral or rock ranging from the size of a finger joint up to the size of a human head;

blocks: coral rocks larger than a human head but still not attached or cemented to the bottom.

rock: the solid rock of the reef.

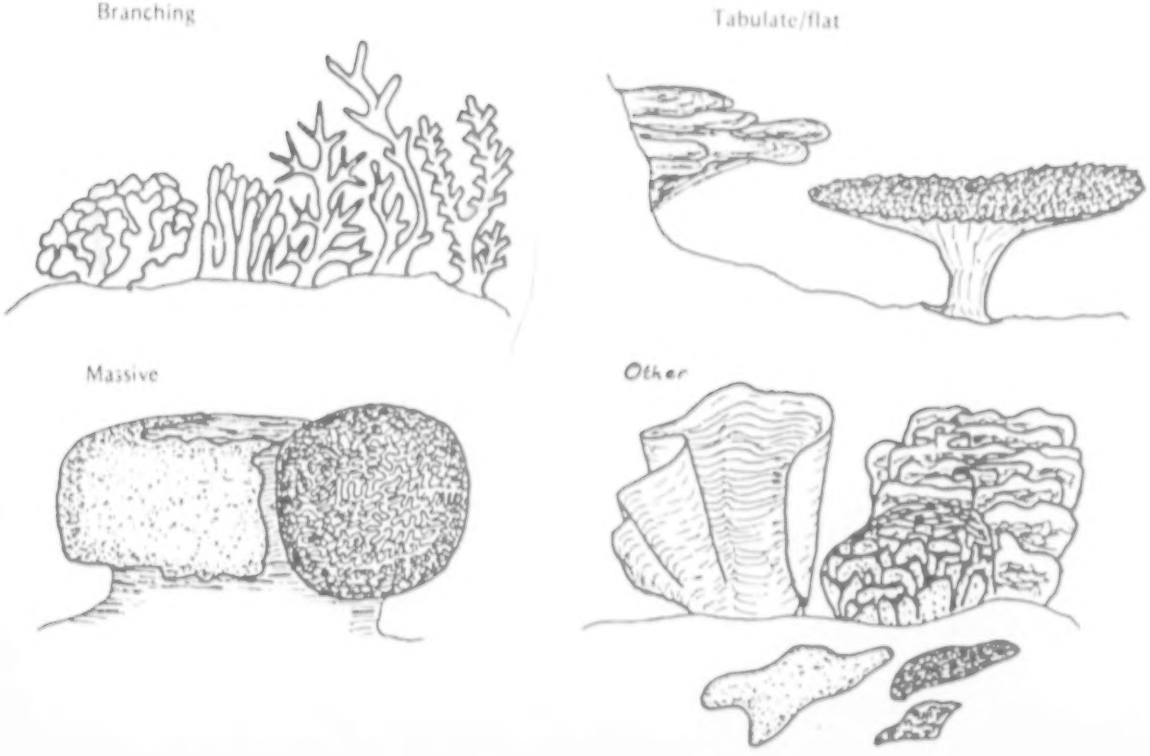
Each group should estimate and record the percent cover of each of these types of bottom in the circle. The sum of all the types should equal 100%.

5. The second measure is the amount of bottom covered by major biological groups attached to it. The following categories are distinguished:

live hard corals: these corals feel hard because they have a stony skeleton, and are covered by the tiny animals or polyps that live in holes in the skeleton. The holes may be like tiny craters, pin-holes or valleys and are often rough or sharp around the edge. The corals generally look "clean" and are coloured by the animals tissues. They may be of many shapes: big like boulders, small and branched like bushes, or just a crust on the rock. Live hard corals can be subdivided into 4 major forms: **branching, massive, tabulate/flat,** and **other.** (Figure 35)

FIGURE 35

FORMS OF LIVE HARD CORALS



soft corals and sponges: these may have shapes like corals but they are soft to the touch, sometimes slimy or rough and flexible. Some are very brightly coloured. If there are waves, they may move back and forth.

dead standing coral: when hard coral dies, only the white skeleton is left. Fuzzy plants may then grow on it, giving it a dirty brown, blackish, pinkish or reddish colour, but the outline of the coral skeleton is still very clear and it is still standing in the place where it grew. This is dead standing coral, and it has not died too long ago, showing that something has killed corals on the reef. Eventually the skeleton is broken up, eroded or overgrown by other things and becomes part of the reef rock and rubble, and is no longer dead standing coral. if it is not clear that something is dead standing coral, do not count it.

crustose corallines: these are plants (algae) that have a stony skeleton-like corals and grow as a crust on dead parts of the reef, cementing it together. They range from dark pink or purple to whitish-yellow in colour (grey in deeper water) and have a smooth or lumpy surface like spilled paint.

marine plants: these include the sea grasses and seaweeds and can range from a cushion-like turf to big floppy plants. They may begreen, red or brown in colour. Only plants or clumps of plants large enough to grab hold of should be counted; fine algal films and fuzzes are too difficult to measure.

Each student group should estimate the percent coverage of each of these categories. The difference between the total of these percentages and 100% is the part of the reef without any obvious organisms.

7. The class should discuss the results of the survey in terms of the importance of each type of bottom to the construction or erosion of the reef. The coverage of organisms should be related to reef processes such as reef construction, food production, and the creation of shelter for fish and other important animals. A low coverage of live hard corals and coralline algae shows a reef that is no longer growing. A significant amount of dead standing coral is a sign that the reef has been damaged recently.

Teaching hints

Care should be taken that the class does the least possible damage to the reef. Walking on a reef, standing on or grabbing corals can cause significant breakage of fragile species. Swimming over the reef is less damaging, if that is possible.

Follow up

This activity can be repeated over time to observe changes in the reef, or used to compare different reef areas. See activities 32 (Monitoring) and 33 (Environmental Impact).

Reference

This is a simplified version of techniques described in the Coral Reef Monitoring Handbook, United Nations Environment Programme, Reference Methods for Marine Pollution Studies No. 25 (1984), available from UNEP OCA/PAC, P.O. Box 30552, Nairobi, Kenya. The whole handbook is suitable for use at secondary school level.

8. TRANSLOCATING MOLLUSCS

(A, C, D; a; III)

Objectives

1. to experiment with factors controlling animal distributions.
2. to learn the value of experimental approaches to ecological research.

Curriculum links

Animal distributions, limiting factors, population dynamics.

Preparation

Habitat:	any intertidal area (rocky shore, sand flat, mangrove trunk) where a species of mobile mollusc, such as a snail or periwinkle, is common in a narrow band or zone.
Time required:	two visits of 2 hours each one to two weeks apart.
Teacher:	select an area where a common local intertidal mollusc is suitable for the experiment. If possible, obtain some information on the ecology of the species concerned (food, predators, resistance to exposure, preferred ecological conditions) to use in explaining the experimental results to the class. Verify that the marking technique used to identify the snails is sufficiently persistent that loss of marks does not affect the result.
Students:	should understand the idea of an experiment and a control.
Equipment:	tools for collecting molluscs (shovel, sieve or knife may be necessary depending on the species and habitat) Fast-drying waterproof paint, nail polish, or permanent marking pens (3 colours) 3 stakes or other means of marking the experimental site.

9. CORAL REEF SURFACE AREA

(A, C; I, II)

Objectives

1. to observe the three-dimensional structure of a coral reef.
2. to relate the structure to environmental factors.
3. to learn the functional importance of surface area.
4. to understand the choices involved in different life-form strategies.

Curriculum links

Functional morphology, community structure, photosynthesis.

Preparation

Habitat: choose a healthy coral reef with as much living coral and vertical relief as possible.

Time required: 1 - 5 hours on the reef and 1 - 5 hours in the classroom.

Teacher: if the students do not have a background in solid geometry, they will need to be provided with the formulae for calculating the surface of such forms as a circle, triangle, rectangle, sphere, cone, and cylinder.

Students: some ability in mathematical operations such as calculating the surface of a sphere or cylinder.

Equipment: measuring tapes, metre sticks, rulers
40 m of rope or cord marked at 10 m intervals
4 stakes or rods
field notebooks and pencils
calculators

Strategy

1. The surface of an organism has great functional significance. It determines the amount of light a plant absorbs, the uptake or loss of gases, water, nutrients, and heat, and then drag that must be resisted in a wind or current, among other things. An ecosystem such as a coral reef also creates surface as it becomes richer. In this activity students will observe how much surface area a coral reef can make, as a basis for understanding its ecological signfiicance.

2. Select a healthy coral reef area with many corals. Mark off a quadrat (square) 10 m on a side with the rope and stakes. Horizontally this quadrat covers 100 square metres.

Strategy

Question: Can an organism survive outside its usual zone of distribution?

1. Select a site where the mollusc is common in one zone but not in adjacent areas above or below the zone. Collect 60 to 150 of them, divide them into 3 groups of 20 to 50, make sure the shells are clean and dry, and mark those of each group with paint of a different colour. The sample size chosen will depend on the numbers of animals available and the time needed to apply the marks.

2. Place three markers (paint can be used on hard surfaces, stakes in soft substrates), at the place where the molluscs were collected, and in zones seaward and landward of that zone (but not too far away) where they do not occur.

3. Release one group of 20 - 50 (the control) in the immediate vicinity of the marker where they were collected, and the other groups around the other markers. Position them so that they can reattach or rebury themselves if possible.

4. Return a few days or a week later, examine the surface or sieve the sand around each marker, and count the number of organisms in each group that can still be found.

5. The numbers of surviving translocated organisms should be compared to the number of control organisms (returned to their original location). What percentage of marked organisms can still be found? How far have the marked organisms moved? Have any transplanted organisms returned to their normal zone?

6. Discuss with the class the environmental factors that could explain the results observed, and possible interpretations in terms of movement, mortality rate and population size.

3. Measure the size of the gross relief of the bottom, such as the size of ridges or depressions, and prepare a sketch map of the quadrat, noting the dimensions measured.

4. List or note on the map the numbers and sizes of the major corals. For each type of coral, measure its dimensions (height, diameter, circumference, number of branches, diameter of branches, distance between points of branching, etc.). Other fixed animals should be counted and their size recorded.

5. Make similar counts and measurements for significant plants in the quadrat. Measure the approximate area covered by plants, the height of the plants, the number of blades or fronds or their average density per unit area, and the size or diameter of the blades.

6. These measurements will be used in the classroom to reconstruct the quadrat with simple geometric forms that approximate to the forms observed on the reef, but for which it is easy to calculate the surface area. For instance, ridges on the reef may resemble a corrugated surface made up of half cylinders; a massive coral may be approximated by a hemisphere of the same dimensions; a branching coral by a set of cylinders branching in the same way (Figure 36 , page 94). The pupils can use their imaginations to recreate theorganisms observed using geometric forms, and then calculate the surface area of the forms. First calculate the surface of each type of coral or other organism at the average size observed, then multiply by the number observed to get the contribution of that organism to the total surface of the quadrat. The total area of the reef surface plus the corals and other organisms gives a surface area index, which can be expressed in square metres of surface for each horizontal square metre of bottom. A reef with many corals might have a surface area index of 15, so the 100 square metre quadrat may have 1500 square metres of functional surface. A moderately well developed reef might have half of that, and a poor reef an index of less than 3.

7. Discuss the importance of the surface observed for such reef processes as absorbing light for photosynthesis, feeding on plankton, exchanging nutrients and waste products with the water, resisting storm waves, etc.

Teaching hints

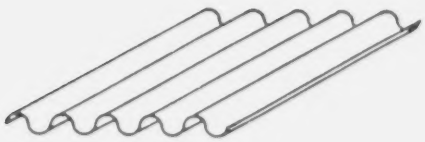
This activity will be particularly meaningful if it follows the study of photosynthesis or other biological processes for which surface is important.

FIGURE 36

GEOMETRIC FORMS APPROXIMATING REEF SURFACES

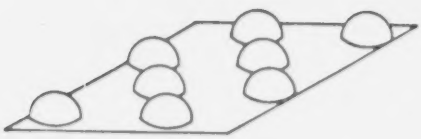
THEORETICAL REEF SURFACE

SCALE I — GROSS REEF MORPHOLOGY

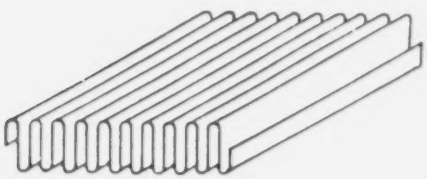


RIDGES 1 M HIGH AND
1 M APART

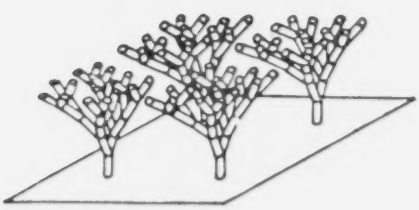
SCALE II — INDIVIDUAL CORALS



25% MASSIVE AND CRUSTOSE
.25 M DIAMETER
HEMISPHERES, 8 PER M^2



25% PLATE-LIKE
RIDGES .25 M HIGH, .05 M
WIDE, AND .05 M APART



25% BRANCHED
CYLINDERS .5 M HIGH, .05 M
DIAMETER, BRANCHED EACH
.1 M, SPACED .1 M AT TIPS

25% NO RELIEF AT SCALE II

SCALE III — CORAL OR ROCK SURFACE

60% CORAL SURFACE
POLYP CUP CONICAL 10 MM
DIAMETER, 5 MM DEEP, WITH
20 SEMICIRCULAR PLATES
2.5 MM HIGH

40% NO RELIEF AT SCALE III

Follow up

It is possible to use this activity to compare different reef zones, reefs exposed to different environmental conditions (such as windward and leeward reefs, or exposed and sheltered reefs). or undisturbed reefs and those that have been damaged by human activities.

Reference

A more complete explanation of surface area estimations on coral reefs can be found in: Dahl, A. L. Surface area in ecological analysis: quantification of benthic coral-reef algae, Marine Biology, Volume 23, 1973, pages 239 - 249.

10. REPLANTING MANGROVES

(C; a, b: III, IV)

Objectives

1. to learn the techniques for replanting mangroves.
2. to see how to experiment with the re-establishment of more productive or natural communities.
3. to appreciate the value of restoring exploited or damaged ecosystems.

Curriculum links

Mangroves, reforestation, restoration of degraded ecosystems, environmental management.

Preparation

Habitat: a former mangrove area where the trees have been damaged or removed but suitable shallow muddy habitat exists.

Time required: 2 - 3 hours of field work plus occasional subsequent visits to observe the results.

Teacher: select a suitable coastal area in need of reforestation with mangroves. Be certain that the conditions that caused the loss of mangroves have been corrected. Identify also an existing mangrove area from which propagules (seedlings) can be collected at a stage suitable for planting. Choose a low tide period when the site to be planted is accessible.

Students: an explanation of mangrove reproduction and dispersal should be given before the field activity.

Equipment: knife for collecting propagules
buckets or plastic bags
small shovels or trowels.

Strategy

Question: How can a mangrove forest be re-created where it has been damaged or lost?

1. Construction, pollution or exploitation can damage or destroy a natural ecosystem that may be valuable for man. We cannot go on destroying nature for ever, so we must learn how to help natural systems grow back orrecover after the damaging influences have been removed. Not much is known about restoring damaged marine ecosystems, but mangrove forests have been replanted in many places. Since mangroves are important for coastal protection, and for the food and shelter they provide many commercially important species, they should be restored whenever possible. This activity will show how to do it.

2. Study your local mangrove community to see the conditions under which each species lives (Activity 3, page 76). Observe the propagules being produced on the trees, and the young seedlings that have fallen, found a suitable site, and started to grow. Measure how far apart the adult trees are spaced.

3. Study the damaged area to be replanted in the same way to identify the parts of the area with conditions appropriate for each species.

4. Collect enough mature propagules to cover the area to be planted at slightly more than the same density as in the natural area (to allow for some mortality).

5. Mark out the spacing in the area to be planted, and plant the propagules at about the same depth as that observed in nature. Be certain that each species is planted in its preferred conditions. Count the number of propagules planted and measure their heights at planting.

6. Return at intervals of 2 to 4 weeks to see how the new seedlings are doing. Count and measure the heights of the survivors.

7. Make graphs of the results, including the number of seedlings surviving over time, and the growth (average height) of the seedlings over time.

Teaching hints

Mangroves reproduce by producing propagules, which are basically seeds that sprout and start growing on the tree, so that when they drop they can root quickly in the mud and not be carried away to sea. Learn to recognise when the propagules are ready for planting.

If many of the seedlings are lost soon after planting, check that conditions in the area are suitable. A storm that washes away new plants is a hazard that perhaps cannot be totally avoided, but waves and erosion are also a danger.

Starting the seedlings in a protected "nursery" area and then transplanting them once the roots have started to develop may improve their holding power in an exposed site. If the seedlings die soon after planting, the problem may be pollution or unsuitable salinity (construction frequently changes drainage patterns, killing mangroves). If the planting activity fails, it will at least illustrate how difficult it is to recreate natural areas.

Follow up

A successful mangrove planting project can be a good demonstration for more widespread environmental restoration by local communities or governments.

If this becomes an annual class project, the class can also visit sites planted in previous years and compare them with undisturbed natural areas to observe the regeneration of the ecosystem.

11. TROPHIC LEVELS

(B; a; I)

Objectives

1. to learn the different tophic levels.
2. to observe which local organisms belong to each trophic level.
3. to understand the prey-predator relations between organisms.

Curriculum links

Trophic levels, food chains, biomass and energy flow, predator-prey relations, feeding adaptations.

Preparation

Habitat:	any intertidal habitat, preferably with attached plants because primary production based on filter-feeding on plankton is more difficult for pupils to visualise.
Time required:	2 hours in the field plus 1 hour class discussion.
Teacher:	learn the feeding habits of local organisms. If time is limited, prepare a list of organisms, possibly illustrated with simple drawings, which pupils can use to note their observations on trophic levels.
Students:	should understand the different trophic levels (primary producers, herbivores, carnivores, omnivores, detritus feeders, decomposers) and the concept of food chains or food webs.
Equipment:	field notebooks and pencils knife or prying tool magnifying glass or hand lens face masks for underwater observations dissecting tools (small knife and forceps) dissecting microscope (optional)

Strategy

Question: What are the plants and animals that make up the coastal food web?

1. Visit the sea shore at a low tide when as many organisms as possible are accessible.

2. Each organism should be observed closely, and on the basis of its appearance and morphology (plant or animal, fixed or motile, type of mouth parts, location and observed behaviour, etc.) the students should assign it to an appropriate trophic group:

   - primary producers: plants and animals with symbiotic algae;
   - herbivores;
   - carnivores (can have several levels)
   - detritus feeders and decomposers

   Note that some organisms may belong to more than one groups.

3. Probable prey organisms or sources of food should be identified.

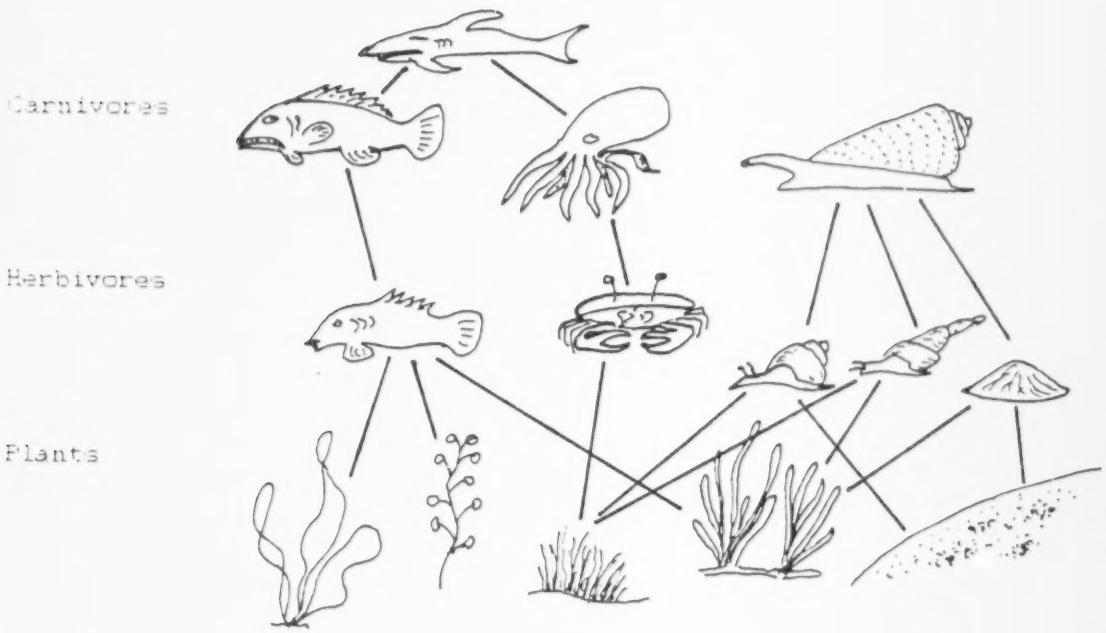
4. If the food of an organism is not apparent, the organism can be collected for examination in the classroom or laboratory using a hand lens or dissecting microscope. Mouth parts can be dissected, and stomach contents examined under a microscope (if available).

5. Any information the students may have on higher levels of the food chain, such as the fish they eat at home, and what those fish eat, can be added to their field observations.

6. On the basis of these observations, the class should construct a food chain or food web for the coastal community they have studied. (Figure 37)

FIGURE 37

EXAMPLE OF A MARINE FOOD WEB



Follow up

This activity can be followed by Activity 24 (page 126) on feeding adapatations.

12. SEAGRASS TURNOVER

(B, C; d; III)

Objectives

1. to observe primary production and loss (turnover) in seagrasses;
2. to use a field experiment to determine change over time.
3. to appreciate the importance of seagrasses in food production in coastal waters.

Curriculum links

Primary productivity, herbivores, detritus, food chains.

Preparation

Habitat:	shallow bottom with seagrass bed accessible to students.
Time required:	two visits of 1 - 2 hours each one week apart.
Teacher:	no special preparation required.
Students:	introduction to the ecological importance of seagrasses.
Equipment:	stiff wire stakes waterproof tags marked with group numbers or student names (can be cut from plastic food containers) 30 cm ruler or measured strings with loop at one end cheap staplers, paper punches or scissors (will rust) field notebooks or waterproof slates and pencils

Strategy

Question: How much food is produced by seagrasses?

1. Each student or small student group should choose a small area of dense seagrass and mark it with a stiff wire stake and attached plastic label driven into the sand. These markers should be left in place for the return visit.

2. The area to be measured is determined by turning the ruler or measured string around the marker. A 30 cm radius is good, but this can be increased or decreased depending on the density of seagrass blades (leaves). There should be 20 to 50 blades in the area measured.

3. Every seagrass blade touched at its base by the string should be counted.

4. All the blades that are counted should be marked in the middle, either with a staple (from an ordinary paper stapler), by punching a small hole in the blade with a paper punch, or by cutting a small notch in one edge with scissors. This will allow the old leaves to be identified on the return visit.

5. The class should return about 1 week later and relocate their markers.

6. Using the same string, they should count both the number of marked seagrass blades remaining and the number of new leaves without marks.

7. From these data, the density of seagrass leaves should be calculated. The number of new leaves is a measure of primary productivity (new food being made). The number of leaves originally marked minus the number of marked leaves remaining after 1 week gives the number of leaves grazed off by herbivores or lost as detritus to supply the detritus food chain.

8. Discuss the significance of the class results for the ecology of the area.

13. MANGROVE LEAF PRODUCTION

(B, A; II)

Objectives

1. to measure the production of organic matter by mangroves.
2. to use a simple quantitative field technqiue.
3. to appreciate the importance of mangrove productivity to coastal ecosystems.

Curriculum links

Mangroves, primary productivity, food chains, detritus, critical habitats.

Preparation

Habitat: any mangrove forest or coastal fringe.

Time required: 1 - 2 hours of initial field work followed by daily visits and 1 hour of laboratory work for the three or four succeeding days.

Teacher: select an accessible mangrove area with a reasonably high canopy density and leaf fall rate, in an area where the leaf traps will not be disturbed.

Students: an understanding of the mangrove detrital food chain.

Equipment: pieces of cloth 1 m x 1 m (such as unbleached muslin)
strong string or cord
laboratory balance or scales (accurate to 0.1 g)
drying oven (if available)

Strategy

Question: How much food is made by mangroves?

1. Mangrove leaves are the beginning of an important detrital food chain in the coastal environment, often extending to lagoons or coastal waters beyond the mangrove forest itself. Important commercial species may depend for part of their life cycle on food energy provided by decaying bits of mangrove leaf and the bacteria that feed on them.

2. To measure the amount of leaf material produced by mangroves, make leaf traps under the mangrove trees. Take 1 square metre of cloth, tie a strong string to each corner, and stretch it horizontally under the mangrove canopy by tying the strings to appropriate trunks or branches. Be sure the traps are high enough that they will not get wet at high tide. Place a rock or other weight in the centre of the trap to give it a funnel shape and to keep the wind from shaking the leaves out of it. Note the location of each trap and the species of mangroves over it. If possible make several leaf traps under each important mangrove species.

3. Return each day for 2 or 3 days (or at whatever interval is practical), remove all the leaves form each trap, put them in separate plastic bags labelled as to the trap they come from, and return them to the laboratory.

4. Dry the leaves to constant dryness (in a drying oven or other warmed place if possible), then weigh them to determine the total organic matter.

5. Average the different sample weights from each trap to determine the organic matter production per square metre per day for each mangrove species measured.

6. During the field visits, observe the mangrove leaves and fragments in the water, as well as any animals that may be feeding on them.

7. Remove the traps at the end of the study.

8. Discuss the importance of mangrove primary productivity. Estimate how much food is produced by the whole mangrove forest by multiplying the production per square metre by the area of the forest.

14. ANIMAL FEEDING

(B, D; a; I, III)

Objectives

1. to observe animal feeding behaviour;
2. to examine what animlas have eaten;
3. to understand how a food chain is determined;
4. to appreciate the relationship between animals important to man and their food supplies.

Curriculum links

Animal behaviour, food chains, herbivores, carnivores, fisheries management.

Preparation

Habitat: any coastal marine habitat where animals can be obtained.

Time required: 2 - 3 hours for each part, divided between field and laboratory activities.

Teacher: select the most appropriate local organisms, preferably macro-herbivores or carnivores; avoid colonial animlas and filter feeders. Try out the exercises in advance to adapt them to local conditions. A familiarity with the anatomy of marine organisms will be useful for the dissections.

Students: introduce concepts of feeding behaviour and adaptations in marine animals before the field activity

Equipment: buckets and other collecting gear
dissecting instruments (scapels or razor blades, pins, forceps, etc.)
aquaria (or large glass jars or plastic basins)
hand lens or dissecting microscope (optional)
microbalance (optional)

Strategy

Question: What and how much do animals eat?

1. Visit a coastal area and collect some large benthic animals or catch some fish that are easy to dissect. Note any examples of feeding behaviour observed, and the environment and possible food sources for the animals collected.

2. Remove the stomach contents of the animals and weigh them (if possible) to see how much the animals have eaten.

3. Examine the stomach contents with a hand lens, dissecting microscope and/or compound microscope to try to identify what the animals have eaten. Compare the fragments observed with what was seen in the animal's environment.

4. Look at the animal's mouth parts. Are they adapted for sucking, biting, scraping, filtering, or catching and swallowing whole?

5. Put an herbivorous animal in an aquarium in conditions that resemble its natural habitat. Be sure that the aquarium conditions will not affect the animal's feeding behaviour. Select a plant that is a known food of the animal. Choose one or more pieces that are easy to quantify. Weigh them, trace their outline if they are flat, or draw them to record the initial amount of plant material. Put them into the aquarium with the animal. After 1 or 2 days reweigh or retrace the plant pieces to see how much has been eaten. Is there obvious evidence of grazing damage?

6. Discuss with the class the significance of their observations. If the animals studied are important in local fisheries, how can a knowledge of their food supply help in fisheries management?

Teaching hints

If it is difficult to obtain animals on the shore, fish for dissection might be obtainable (uncleaned) from a local fisherman.

Ingested fragments of animals and plants are often hard to identify. Spend some time examining the gut contents of local organisms before introducing this as a class project to prevent more questions being raised than can be answeered at the time.

Follow up

Feeding studies can be related to the functional morphologies involved in feeding (See Activity 24, Feeding Adaptations, page

15. FEEDING PREFERENCES

(B, D; III)

Objectives

1. to observe that marine animals have feeding preferences;
2. to learn to test observations experimentally.

Curriculum links

Animal behaviour, feeding, herbivores, animal-plant interactions.

Preparation

Habitat: any coastal area with hard substrate where herbivorous molluscs or similar animals are comon.

Time required: 1 - 2 hours in the field, plus observations in the laboratory or classroom.

Teacher: select a common local marine snail or similar herbivore that is easy to keep several days in an aquarium. Test the organism selected to be sure it will feed in the aquarium befope doing the class experiment. Note that some high-intertidal snails may prefer to be wet but not covered by water.

Students: previous work on other aspects of animal feeding is advisable.

Equipment: marine aquarium
field collecting materials (bucket, knife, etc.)

Strategy

Question: Do marine animals have feeding preferences?

1. Visit a coastal intertidal area where the mollusc selected is common. Observe all the potential food plants available in its habitat.

2. Note where the mollusc is found. Does it occur on or near some plants more than others? Are there signs of feeding on certain plants?

3. If the molluscs are inactive because the surface is dry, try wetting them with seawater. Do they begin feeding after a few minutes wetted?

4. Collect a number of molluscs and samples of the different available food plants and surfaces in their habitat. Return them carefully to the laboratory.

5. Put the molluscs in a marine aquarium with samples of the available food plants attached to rocks or weighed down to make them accessible. Observe them over a few days and record on which plants the molluscs spend the most time feeding. Note that some animals may only feed at certain times of the day or tide. List the plants in order of feeding preference.

6. Discuss with the class the possible explanations for any feeding preferences observed (plant form, texture, taste, etc.) and the significance of feeding preferences for the ecology of the coastal zone. Also discuss possible effects of the laboratory situation as compared to what might occur normally in the field. How might such differences be identified and quantified?

16. MARINE PLANKTON CULTURE*

(B; b; I)

Objectives

1. to observe the role of plankton as food in aquaculture.
2. to learn how to culture phytoplankton.
3. to understand the usefulness of phytoplankton.
4. to develop skills useful for culturing other microorganisms.

Curriculum links

Micro-organisms, nutrition, aquaculture.

Preparation

Habitat: field excursion to any aquaculture operation or marine laboratory engaged in breeding aquaculture organisms.

Time required: 2 - 3 hours field trip, 2 - 3 hour initial laboratory preparation, and daily observations over 10 days.

Teacher: obtain phytoplankton samples from an aquaculture farm or marine laboratory and establish stock cultures.

Students: an introduction to food chains and aquaculture would be useful.
Divide students into small groups.

---

*Based on an activity devised by Dr. Twee Harmchong.

Equipment:

inexpensive aquaria, glass jars or widemouth bottles
plankton net
phytoplankton: Chlorella, chlamydomonas, Tetraselmis, Dunaliella, etc.
disinfectant: formaldehyde, malachite green, etc.
pond water
fertiliser: ammonium sulphate, composted manure, or sterilised soil
microscope slides and cover slips
eye dropper
small air pump (optional)
compound microscope (optional)

Strategy

Question: What do aquaculture organisms eat and how is their food raised?

1. Coastal aquaculture of marine and brackish water animals (prawns, shrimp, clams, oysters, fishes) is spreading rapidly in many countries. The larvae of these marine animals generally eat living phytoplankton (microalgae), which must be cultured in quantity if such aquaculture is to succeed.

2. Students should first visit an aquaculture farm or marine laboratory where they can observe phytoplankton cultures and learn their importance to the organisms being raised.

3. In the classroom or laboratory, each group should prepare a clean aquarium or culture bottle.

4. Disinfect pond water by heating to boiling or by chemical treatment: formaldehyde (37%) 50 ppm for two days, or malachite green 100 ppm for 6 hours. Fill the culture container about $\frac{2}{3}$ full with disenfected water.

5. Add fertiliser: 20 g ammonium sulphate, sterilised soil or sterilised composted manure (approximately 1 kg).

6. Innoculate (add to) the culture container 5 ml of pure stock culture of the phytoplankton.

7. Supply air to the culture container with a small air pump, if available.

8. Place the culture containers in sunlight.

9. Observe the multiplication of the phytoplankton daily for 10 days by recording the colour of the culture water, and checking and recording the density of microalgae under a microscope.

10. Filter the culture at the end of the experiment with the plankton net and observe the quantity of algae produced.

Teaching hints

It is important to avoid any contamination of the phytoplankton cultures by zooplankton.

Be careful that no toxic chemicals or residues get into the cultures.

Phytoplankton cultures can be maintained by harvesting samples and innoculating them into new culture medium.

Follow up

Suggested questions for discussion:

1. What was the difference in density of microalgae between the first and the tenth day?
2. Why did the culture water change colour?
3. What does phytoplankton look like?
4. How many microalgae did you obtain in one litre?
5. Do you believe that microalgae or phytoplankton could be used as human food?
6. What are the reasons for the different precautions and additives?
7. What are the possible sources of any problems with the cultures?

17. CORAL GROWTH RATES

(C; d, a; II, III)

Objectives

1. to observe rates of growth and skeletal accumulation in corals.
2. to learn the importance of precise measurement and data recording in the field.
3. to see species differences.
4. to appreciate the importance of avoiding damage to corals.

Curriculum links

Coral reefs, calcification, growth rates.

Preparation

Habitat — select a field site where different types of corals are common and where access at low tide is not too difficult. A reef flat or shallow back reef area would be suitable.

Time required: — 3 - 4 hours for the first visit and 1 - 2 hours for each subsequent visit. Measurements should be made approximately monthly for at least 3 - 4 months and preferably longer.

Teacher: — if class time is limited, the study area can be marked and the corals to be measured can be identified in advance.

Students: — to be able to recognise different types of corals. Some practice in labelling and measuring techniques using dried corals in the classroom may be useful before the field exercise, as precise measurements are required.

Equipment:

plastic rulers (20 or 30 cm long, graduated in mm)
cloth measuring tapes (such as used by tailors)
string or fishline
small rigid plastic tags with punched holes, numbered with pencil or waterproof ink (may be cut out of think plastic food containers)
2 steel rods (reinforcing steel) about 1 m long
heavy hammer
field notebooks or slates and pencils
facemasks or goggles (optional)

Strategy

Question: How fast do corals grow?

1. The rate of coral growth helps determine the rate of reef growth as well as the speed with which a coral reef can recover from damage. This activity involves measuring different corals to see how fast they grow.

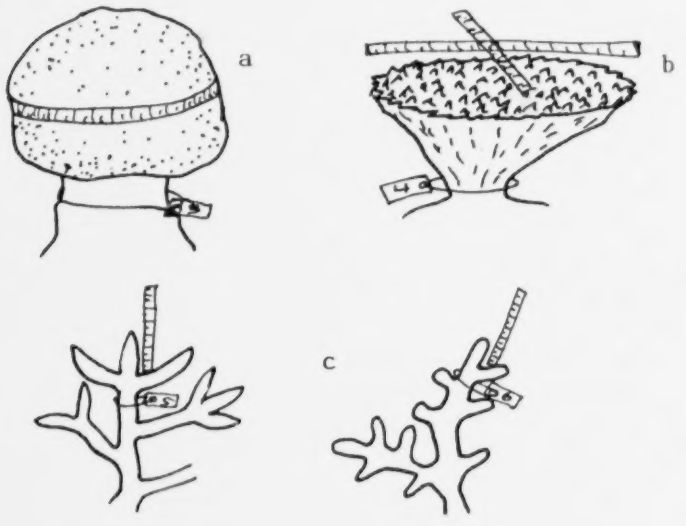
2. The study area should be marked by driving steel rods into the reef so that it can be found again easily.

3. Working singly or in small groups, pupils should select at least one massive or table coral and two types of branching corals tomeasure. The massive or table coral should be of moderate size (20 - 40 cm in diameter) and as close to round as possible. One branching coral should be of the staghorm type (Acropora cervicornis, A. formosa, etc.) known to grow rapidly.

4. Each coral should be labelled by tying a plastic label firmly around the base (massive/table corals), or loosely around a branch just below the ultimate branch. (Figure 38, page 110)

FIGURE 38

LABELLING AND MEASURING CORALS FOR GROWTH



5. A sketch map should be made showing the locations and numbers of the labelled corals related to the markers.

6. The size of the corals is measured as follows:

a **massive coral:** measure the largest circumference horizontally with a cloth tape measure;

b **table coral:** measure the largest and smallest diameters across the top with a cloth tape measure or ruler;

c **branching coral:** measure the length of the tips from the point of branching just above the label (if there is more than one tip, measure all the tips. (Figure 38)

7. These measurements should be recorded in a field notebook together with the date, the label number, and a sketch of each coral showing what it looks like and how it was measured.

8. The corals should be remeasured in exactly the same way approximately once a month for at least 3 or 4 months. If new branches form on the branching corals this should be noted in the sketches, but measurements should continue to be made from the original starting point.

9. The amount of growth of each coral in circumference, diameter or length should be calculated by subtracting the first from the last measurements. Monthly growth can also be plotted as a graph. The class results for each species can be analysed for statistical significance if desired.

Teaching hints

1. Care must be taken not to break or injure the corals while labelling or measuring them.

2. Since corals grow slowly, considerable precision will be needed in measuring to detect growth. Massive corals in particular may show little or no measurable growth in the time available. This in itself is signfiicant for the pupils to understand.

3. Unexpected results such as decreasing rather than increasing measurements should be discussed in terms of possible sources of error such as damage to the coral or variability in the measuring technique. The amount of variability can be tested through repeated measurements of dried corals in the laboratory.
4. If labels are lost they should only be replaced and measurements continued if there is no doubt as to their precise position, particularly for branching corals.

Follow up

Coral growth rates can be discussed in the class with reference to the size and thus the age of corals observed on the reef, and the rates of coral regeneration after damage by destructive fishing techniques, etc. They can also be related to the rate of growth of the reef itself, and the time it has taken for some reefs to become hundreds of metres thick.

18. ANIMAL POPULATION SIZES

(A, C; a, c; II)

Objectives

1. to determine the numbers of some common benthic (= living on the sea floor) marine animals.
2. to observe the relationship between animal populations, environmental factors and human impact.

Curriculum links

Animal populations, fisheries, environmental impact.

Preparation

Habitat: a shallow coral reef is best, but a rocky shore, mangrove area or mud flat with adequate animals can also be used.

Time required: 2 - 3 hours in the field.

Teacher: select a site with as many interesting benthic animals as possible. The activity and its interpretation will have to be modified somewhat depending on the type of habitat chosen.

Students: the types of animals to be counted should be studied before the field activity. (If the students have experience in data processing, the results can be processed on a micro-computer with suitable programmes prepared before the fieldwork.)

Equipment: a rope and stakes, or square or round quadrat frame, for defining areas within which animals are to be counted
shovels and sieves if the animals are in sediments
field notebooks or slates and pencils
micro-computer (optional)

Strategy

Question: How many animals are there per unit of area.

1. Quantitative measures of animal populations per unit area are often useful in ecology. This activity involves conting some obvious or important animals in the coastal marine environment. The animals counted, the techniques used, and the areas sampled will all depend on the tpye of habitat. The following suggestions for some typical habitats will need to be adapted to local conditions.

2. Lay out one or more areas to be sampled:

   coral reef: drive a stake into the reef, and define a 50 square metre area in a circle around the stake using a rope 4 metres long with loops at both ends.

   sandy/muddy bottom: use one square metre quadrats, digging up and sieving the sediment to a depth of 20 - 30 cm.

   rocky shore: the quadrat size defined by a quadrat frame or rope and stakes can vary from 1 square metre to 50 square metres, depending on the animals to be counted.

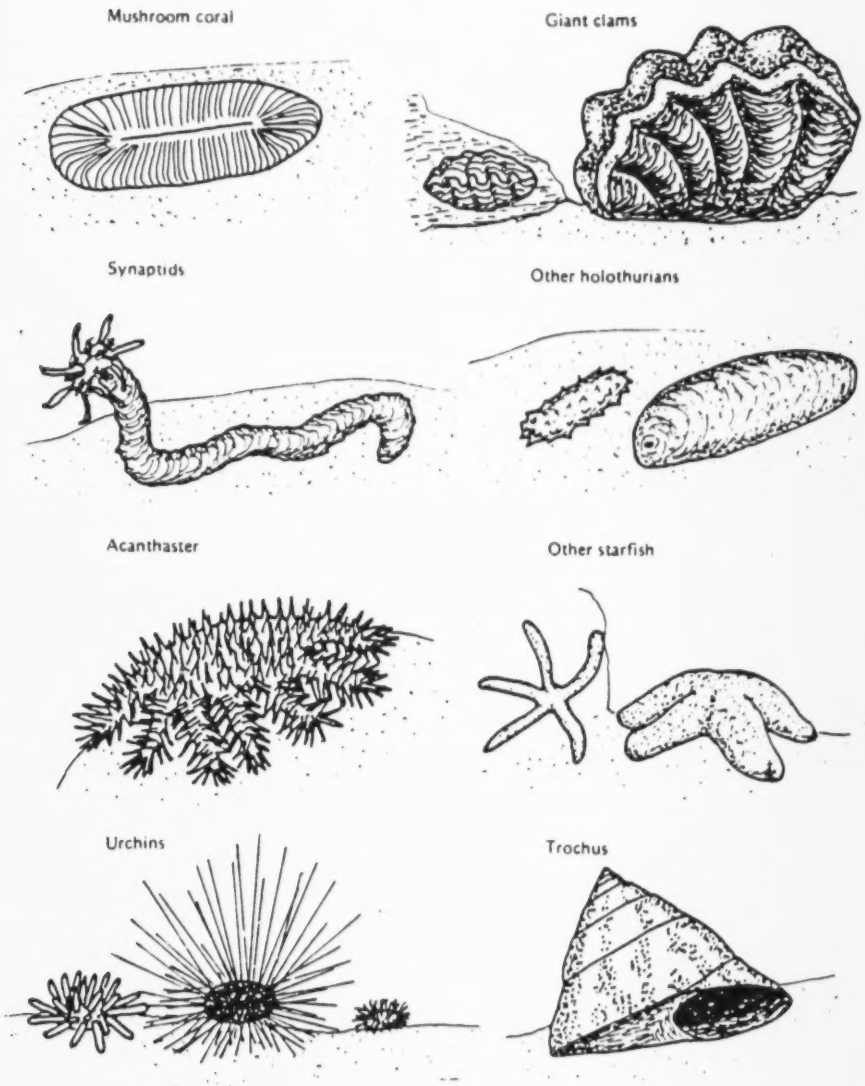
3. Count the numbers of common or obvious animals in the sample area, especially those with indicator value:

   coral reef: mushroom corals (Fungia spp.) are among the first things to be picked up by tourists; giant clams are often subject to heavy fishing pressure; synaptid holothurians may incrase in number with heavy organic pollution; other

holothurians are sometimes fished for food as beche-de-mer; Acanthaster (Crown-of-thorns) starfish may become numerous and damage the reef; other starfish are often obvious and colourful; sea urchins may go through big population changes; large shellfish like Trochus are often collected for food or export. (Figure 39)

FIGURE 39

CORAL REEF ANIMALS TO BE COUNTED



sandy/muddy bottom: bivalve molluscs may be important food organisms; crabs may be numerous and ecologically important.

rocky shore: mussels, oysters and snails may be common food organisms; barnacles may be ecologically important; starfish are important predators, etc.

4. If several samples are taken, compare the results between samples as related to zonation, environmental or human factors.

5. Process the data using tests of statistical significance and presentating the results in graphic form. (Use a micro-computer if available.)

Teaching hints

Select the few animals tha best illustrate topics in the curiculum, or that are important in the local economy.

Follow up

Animal counts lend themselves to monitoring and environmental impact studies (Activities 32 and 33 pages 145and 147) and analysis of the effects of overfishing (Activity 30 page 140). They can also be correlated with environmental parameters.

Reference

Based in part on the Coral Reef Monitoring Handbook, United Nations Environment Programme, Reference Methods for Marine Pollution Studies No. 25 (1984), available from UNEP OCA/PAC, P.O. Box 30552, Nairobi, Kenya.

19. SETTLEMENT AND SUCCESSION

(A, c: a; III)

Objectives

1. to observe the colonization of newly-exposed surfaces.
2. to follow the succession of colonising species over time.
3. to test whether settlement and succession vary in different environments.

Curriculum links

Population dynamics, colonization, succession, competition.

Preparation

Habitat: any intertidal or shallow subtidal area without excessive wave action or sedimentation, preferably with abundant benthic life.

Time required: 1 hour in the field to set up the experiment, plus return visits approximately monthly for three months. The materials collected on the return visits will need 1 - 2 hours of study in the laboratory or classroom after each visit.

Teacher: if literature on local fouling organisms is available it will help in the interpretation of class observations. Some experience in the speed of recolonization locally may be needed to adjust the sampling interval to suit local conditions.

Students: an understanding of reproduction, planktonic larval stages, and fouling will reinforce the effectiveness of this unit.

Equipment: concrete blocks, bricks, distinctive rocks or other heavy hard surfaces, brushed clean and well washed; sterilise by boiling or by washing with alcohol if they have been in the marine environment. (3 per small student group)
buckets
dissecting microscope or hand lenses
steel rods, wire or other means of attachment attachment (optional)
compound microscopes (optional alternative)
glass microscope slides (optional)
masking tape or large rubber bands (optional)

Strategy

Question: How does marine life become established on newly available surfaces?

1. Each small group of students should prepare 3 blocks, bricks or other substrates, taking care that they are clean and free of organisms and toxic substances.

2. The blocks are taken to the field site and placed on the bottom. The location of the blocks should be carefully noted so that they can be found again easily. If time allows, sets of blocks can be put in different zones of the intertidal or at different locations.

3. About 1 month later, the class should return to the same site, collect one block from each environment, and bring it back to the laboratory in a bucket.

4. In the laboratory, they should examine the surface of the blocks with a dissecting microscope or hand lens for any newly settled organisms and make notes describing what they see, with the help of any references that may be available.

5. A second block per site is collected and examined about 1 month later, and the third after 3 months. The observations after 1, 2, and 3 months should be compared.

6. If compound microscopes are available, an alternative rapid microfouling study can be done by attaching 4 microscope slides to each brick with masking tape or rubber bands, leaving the centres uncovered. The bricks are put out intertidally or subtidally and one slide is removed each day for 4 days. The slides are transported carefully to the laboratory and examined uner a compound microscope for diatoms and other micro-organisms that have settled on them.

7. The comparative results over time should be discussed in the class. Which organisms appeared first? Did any organisms become less common over time? If so, what replaced them? Were there signs of grazing on the blocks or slides? Where did the settling organisms come from?

8. The class should prepare a diagram or description of settlement and succession on the blocks or slides

9. If blocks or slides were placed at different levels or sites, the patterns of settlement and succession at each site should be compared.

Teaching hints

This activity needs a site where the blocks will not be disturbed by natural or human factors. The blocks should be heavy enough not to be turned over by waves.

If microscope slides are used, a method should be developed for transporting them to the laboratory, preferably still submerged in sea water, while protecting any organisms from being rubbed off the top surface.

20. REEF FISH SURVEY

(A; a, c, d; I)

Objectives

1. to observe the number of fish on a coral reef;
2. to determine the status of reef fish populations.

Curiculum links

Animal populations, coral reef ecology, fishing effects.

Preparation

Habitat: a well-developed coral reef area, such as a reef slope, back reef or lagoon reef, where students can safely swim while counting fish. Clear water with low turbidity is desirable (visibility at least 5 metres).

Time required: 1 - 2 hours in the field.

Teacher: identify sites for the fish surveys that are easily adn safely accessible.

Students: good swimming ability is required for this activity; familiarity with the types of fish to be counted is important.

Equipment:
face masks or goggles
snorkels and swim fins (desirable)
100 metres of floating polypropylene rope or string
field notebook or underwater slate

Strategy

Question: How healthy are the fish populations on the reef?

1. Select an area along the face of the reef or reef slope parallel to the shore, with fish populations typical for the area, and water depths not exceeding 5 metres.

2. Attach one end of the 100 metre rope to a fixed point (coral head, stake or anchor) or have sufficient students holding it to be able to pull a swimmer back to shore. The student making the survey will swim in a straight line for 100metres along the reef by holding the other end of the rope and swimming until it is stretched to its full length. Fish counts will be made for 2.5 metres on either side of the line (width of the area counted: 5 metres; total area: 500 square metres). One count can be made swimming out along the line, and another swimming back to the starting point.

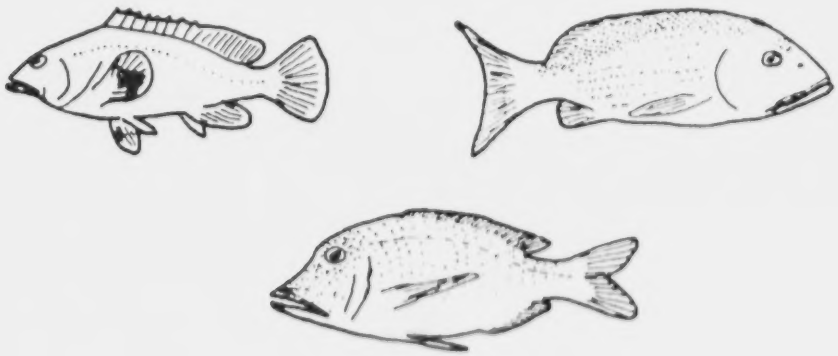
3. Two groups of fish are to be counted (see Figure 40)
Predators: These are larger fish, longer than an outstretched hand, that ea other fish, and include the groupers (Serranidae), the snappers (Lutjanidae) and the emperors (Lethrinidae). (The types of fish expected to be found in a particular area should be ascertained beforehand.) They should be counted first swimming out along the line, as they may be frightened away by a swimmer going by.

Butterfly fish: The butterfly fish or chaetodontids are small brightly coloured reef fish that are quite easy to tell from other fish on the reef. They often swim in pairs and have a special way of biting and sucking around corals. They are not so disturbed by swimmers, tending to stay over the same part of the reef, and so can be counted on the return swim to the starting point.

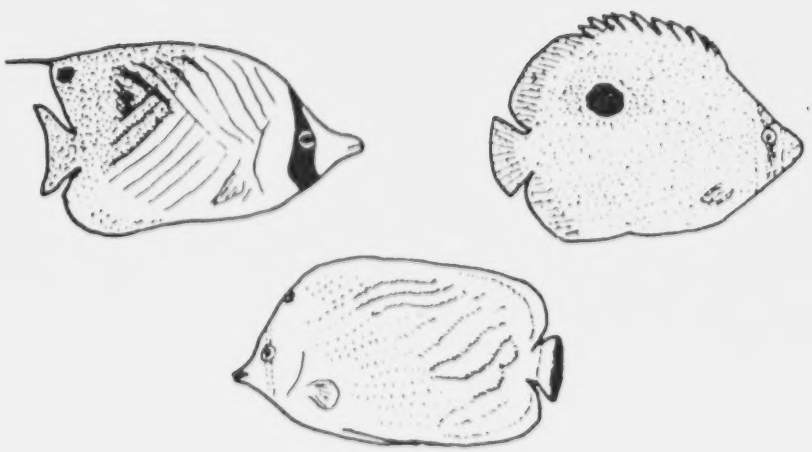
FIGURE 40

REEF FISH TO BE COUNTED

Predators (snappers, groupers, emperors)



Butterfly fish



Fish to be counted (details of shape and colour pattern may vary; not to scale)

4. The swimming student should count the number of predators and the number of butterfly fish in the survey area. Care should be taken not to count the same fish more than once.

5. Other student groups can repeat the survey in adjacent areas for a larger sample of reef fish populations.

6. The significance of the results should be discussed with the class. The numberof larger predatory fish is a good measure of the fishing pressure on the reef. If they are rare or absent, there has probably been overfishing. The small reef fish like the butterfly fish show whether the reef can support many larger fish. Usuallymore than 10 are counted in a 500 square metre survey on a healthy reef. They may decrease as a result of bad fishing methods as when explosives, poisons, small mesh nets or traps are used, or because of aquarium fish collectors. The number of fish can also decrease because something has damaged the coral and other situation that give the fish food and shelter.

Follow up

The reef fish survey method can easily be used to measure overfishing (Activity 30), or environment impact (Activity 33), as well as in monitoring (Activity 32) and conservation surveys (Activity 34).

Reference

This activity is based on a technique described in the Coral Reef Monitoring Handbook, United Nations Environment Programme, Reference Methods for Marine Pollution Studies No. 25 (1984), available from UNEP OCA/PAC, P.O. Box 30552, Nairobi, Kenya. The whole handbook is suitable for use at secondary school level.

21. LOCOMOTION

(A, D; I)

Objectives

1. to observe different types of locomotion in marine animals.
2. to relate functional morphologies to environmental factors.
3. to appreciate the diversity of possible solutions to an evolutionary problem.

Curriculum links

Animal behaviour and adaptations, morphology, evolution.

Prepaation

Habitat: any coastal marine habitat where animals can be observed or collected.

Time required: 2 - 3 hours of field and/or laboratory observations.

Teacher: select the best conditions for observing locomotion in marine animals. If shallow lagoons or tide pools are available, then field observations may be adequate. Otherwise it will be better to catch the animals and put them in aquaria or large glass jars to observe them, either in the field or back in the laboratory.

Students: an introduction to the concept of functional morphology (why organisms have the form they have) is essential.

Equipment: dipnet, knife, scraper, shovel for collecting animals,
glass-bottomed buckets, viewing boxes or face-masks
aquaria or large glass jars with seawater
dissecting microscope (optional)

Strategy

Question: How and why do animals move around?

1. Organisms rarely have features that are not of some use. To appreciate the diversity of life we need to discover why animals are the way they are, and how they have solved the problems of life in their particular environment. This activity looks specifically at some of the kinds of locomotion used by marine animals.

2. Visit an intertidal field site and observe as many animals as possible in their natural environment. Many benthic marine animals move around as larvae, but stay fixed in one place as adults because the moving water brings food to them. Look particularly for those animals that move around as adults. Observe them in the field, and if possible bring some back to watch more closely in the laboratory.

3. Try to observe as many of the following types of animals as possible: fish, starfish, sea urchin or sand dollar, crab, shrimp, an isopod, snail (gastropod), limpet, scallop and worms.

4. For each animal, ask: How does it move around: swimming with fins, a tail or leg paddles; propulsion by squirting water; walking on legs; holding and creeping with muscular foot; pulling with hydraulic feet; pushing or squirming through sand or mud, etc.? Look in detail at the mechanisms for locomotion.

5. Coastal marine animals must be able to move around without being caried away by strong waves or currents. How do the different animals do this: holding on tightly; moving faster than the water, hiding when conditions are bad, etc.?

6. Why does each animal move around in its particular way: to look for food, to escape from predators, to find a mate and reproduce, to avoid bad environmental conditions, etc.?

7. Through class discussion or small working groups, try to define for each animal its strategy for locomotion. For example, the starfish uses hydraulic tube feet to pull itself over the rocks while holding on tightly to avoid being carried away by waves. It seeks food, and can move slowly to escape bad environmental conditions as when it is uncovered at low tide, but it cannot move away quickly from predators.

22. COLORATION

(A, D; I)

Objectives

1. to observe different types of coloration in marine plants and animals.
2. to relate these functional morphologies to environmental factors.
3. to appreciate the diversity of possible solutions to an evolutionary problem.

Curriculum links

Plant and animal adaptations, morphology (form and colour), evolution.

Preparation

Habitat: any coastal marine habitat where plants and animals can be observe or collected.

Time required: 2 - 3 hours of field and/or laboratory observations.

Teacher: select the best conditions for observing the variety of colours in marine plants and animals. Most organisms can be observed in place. For some of the more mobile animals such as fishes, if lagoons or tide pools are available, then field observations may be adequate. Otherwise it will be better to catch them and put them in aquaria or large glass jars to observe them, either in the field or back in the laboratory. However protective coloration will not be as obvious outside of the natural environment.

Students: an introduction to photosynthetic and auxiliary pigments in plants, and to the roles of coloration in animals (why organisms have the colour they have) is necessary.

Equipment: dipnet, knife, scraper, shovel for collecting organisms
glass-bottomed buckets, viewing boxes or facemasks
aquaria or large glass jars with seawater
dissecting microscope (optional)

Strategy

Question: Why are plants and animals coloured the way they are?

1. Even the colours of organisms have a purpose. The many colours of marine plants and animals help them to live better in their environment. This activity looks specifically at the functional roles of coloration used by marine plants and animals.

2. Visit an intertidal field site and observe as many plants and animals as possible in their natural environment. The students should make field notes or drawings of the colours and colour patterns they observe.

3. For the plants, the colours observed should be interpreted in terms of the roles of photosynthetic and auxiliary pigments in absorbing light energy for photosynthesis. Why do marine lants have more such colours than land plants? (Explain in terms of the different spectral composition of light in water.) Is there any evidence of adaptations to protect from too much light (calcification, reflection)?

4. For the animals, look for examples of the following:
   - protective coloration or camouflage to match the background to avoid being seen by predators;
   - colour patterns to permit recognising others of the same species for group behaviour, territoriality or mating;
   - bright obvious colours or patterns to signal that the organism is dangerous or distasteful;
   - patterns or markings to deceive predators as to the size or direction of the organism (location of the head or eye) or to mimic another (dangerous) species.

5. Collect certain animals with protective coloration, and compare their visibility in their natural habitat and in habitats where they do not normally occur. The students should try to put themselves in the position of a predator looking for something to eat.

6. Discuss with the class the selective pressures that could cause such coloration to develop.

Teaching hints

The subject of coloration can first be introduced with classic examples from elsewhere in the world which are often illustrated in books. The students can then look for similar examples in their own environment.

23. PROTECTION

(A, D; I)

Objectives

1. to observe different features providing protection in marine plants and animals.
2. to relate these functional morphologies to environmental factors.
3. to appreciate the diversity of possible solutions to a series of evolutionary problems.

Curriculum links

Form and morphology, adaptation to environmental conditions, animal behaviour, evolution.

Prepaation

Habitat: any coastal intertidal habitat where plants and animals can be observed.

Time required: 2 - 3 hours in the field.

Teacher: prepare in advance a list of the local environmental factors from which intertidal marine organisms must protect themselves. This can also be done as a class exercise.

Students: an introduction to functional morphologies (why organisms have the form they have).

Equipment: dipnet, knife, scraper, shovel for removing or uncovering plants and animals
glass-bottomed buckets, viewing boxes or face-masks

Strategy

Question: How do plants and animals protect themselves in the difficult conditions of the intertidal environment?

1. Organisms must protect themselves from many things in the environment, and many of their features have a protective function. We need to discover how plants and animals have solved the problems of life in their particular environment. This activity looks specifically at some of the kinds of protection used by marine plants and animals.

2. Visit an intertidal field site and observe as many organisms as possible in their natural environment. For each organism, go through the list of factors from which protection is needed, and discuss how the organism has solved the problem of protection. Alternatively, students can observe the organisms and try to decide what their protective features are.

3. Some environmental factors producing protective responses in the tropics are: sunlight, heat, drying out, exposure to rain or freshwater (low salinity), high salinity from evaporation, wave shock and drag in moving water, abrasion by moving sand and rubble, and predation. For each habitat determine which factors are the most apt to be limiting, and how the organisms present protect themselves.

4. Make a list of the kinds of protective features observed. Some common types of protection are: calcified walls and shells (which sometimes can be completely closed or sealed); hard skeletons or cuticles; sharp spines or spicules outside or in the flesh (as in some sponges); flexible joints; tough rubbery texture; water holding capacity inside a shell, between closely packed branches, or inside a sack or spongy tissue; slimy mucus covering; smooth streamlined forms that reduce drag in water; etc. Note that there are oftent rade-offs between different kinds of protection; spines may deter predators but increase drag, a shell may protect from waves but reduce the cooling effect of evaporation. Determine which kinds of protection are most common in different organisms and in different habitats.

Teaching hints

This activity can be organised in different ways depending on the previous studies of the students. Some approaches can be: list all the kinds of protection observed; identify examples of each of the kinds of protection on a list provided by the teacher; describe the kinds of protection used by certain listed plants and animals; define the environmental parameters of certain habitats and identify the protective adaptations of the organisms in them.

Follow up

Experiments can be designed to test the effectiveness of certain protective features, such as the strength of a shell or attachment, the resistance to drying out or to salinity changes, or temperature tolerances (See Activity 26, page 132)

24. FEEDING ADAPTATIONS

(B, D; I)

Objectives

1. to observe different types of feeding adaptations in marine animals.
2. to relate functional morphologies to the constraints imposed by food supply.
3. to appreciate the diversity of possible solutions to an evolutionary problem.

Curriculum links

Feeding, animal behaviour and adaptations, morphology, evolution, symbiosis.

Preparation

Habitat: any coastal marine habitat where animals can be observed or collected.

Time required: 2 - 3 hours of field and/or laboratory observations.

Teacher: select the best conditions for observing feeding activities in marine animals. If shallow lagoons or tide pols are available where animals feed at low tide, then field observations may be adequate. Otherwise it will be better to catch the animals and put them in aquaria or large glass jars to observe them, either in the field or back in the laboratory.

Students: an introduction to types of food and feeding behaviour would be useful before the field activity.

Equipment: dipnet, knife, scraper, shovel for collecting animals
glass-bottomed buckets, viewing boxes or face-masks
aquaria or large glass jars with seawater
dissecting microscope (optional)

Strategy

Question: How do marine animals get their food?

1. All animals have to get food in one way or another. At the same time, the prey species of plants and animals have evolved many defences to aboid being eaten. In the competition to get a share of the limited food available, animals have evolved many ways of feeding. Predators have to find ways of overcoming the defences of their prey, and of specialising or dividing up

the resource to reduce competition. This activity looks specifically at some of the kinds of feeding adaptations used by marine animals.

2. Visit an intertidal field site and observe as many animals as possible in their natural environment. The following categories are some of the feeding adaptations to look for.

3. Some animals simply catch and swallow their prey. They often have large mouths with sharp teeth for holding or taking bites out of prey. Animals that specialise in small prey may have small mouths to match.

4. Other animals sting or paralyse their prey before eating them. They may have many small stinging cells (nematocysts in sea anemones etc.) on their tentacles, or a large poisonous spear or barb. This enables them to eat big or fast animals that would otherwise escape.

5. There are animals, such as snails and sea urchins, that simply scrape or graze their prey off the rock or reef surface. They may have hard beaks for scraping, small teeth or pinchers for nipping or biting, or even a sort of rasp which keeps growing as the end wears out.

6. Others, such as many bivalve molluscus, may simply suck in their food, or even create a flow of water to bring in plankton which they can then filter out.

7. Predators feeding on hard shelled animals can either break the shell, or bore a hole through it, or even slip their stomach through any little space and digest the animals in its own shell.

8. Parasites may simply attach themselves to a host and live on the juices, or even live inside the host's body.

9. Some animals have found the easiest solution of all. They simply grow their food inside their own body, like the corals and giant clams with symbiotic algae; the algae (zooxanthellae) make food from the sun and then release some of it to the host animal in return for fertilisation by some of its waste products. Each one helps the other in what is called a symbiosis.

10. Students can list animals by the types of feeding adaptation they observe, or they can start with a list of feeding adaptations and try to find examples of each type, or be given a list of animals and determine through observation their particular feeding adaptations.

11. Some animals may be easier to observe feeding in an aquarium. Collect both the animals and their preferred food, and place them together where students can observe their feeding behaviour and how their anatomy is adapted to a particular type of feeding.

12. Discuss with the class how different feeding adaptations help to partition the food available in the coastal environment.

25. WATER QUALITY SURVEY

(A, B; c, d; II, IV)

Objectives

1. to survey the water quality of a coastal area.
2. to relate the water quality to other observations on the state of the environment.
3. to understand the effects of man on productive ecosystems.

Curriculum links

Water quality, pollution, environmental impact.

Preparation

Habitat: any coastal marine habitat, preferably with both polluted and unpolluted areas.

Time required: 1 or more field trips of 2 - 3 hours, depending on the length of coastline to be surveyed.

Teacher: collect any existing information on coastal water pollution. The sophistication of the survey will depend on the analytical equipment available.

Students: an understanding of the sources and types of water pollution and their effects on the marine environment.

Equipment:
thermometer
secchi disk (20 cm weighted black and white disk attached in its centre to a rope marked in metres)
tape measure (10 m or more)
metre stikc
sieve (2 mm mesh size)
laboratory balance (optional)
device for measuring salinity (hydrometer, refractometer, or conductivity meter; a crude comparative measure of salinity can be made by evaporating known volumes of seawater and the test samples to dryness and weighing).
see page
field noteboosk and pencils

Strategy

Question: What is the quality of the coastal waters?

1. Most water pollution measurements require sophisticated techniques and equipment. However a few simple indicators can be used to determine the general quality of coastal waters. These measures should be made at regular intervals along the coast, perhaps every half kilometre or kilometre, or at least at several points around each harbour, bay, beach or other coastal feature. (See Chapter 4.)

2. At each survey point, take and record the temperature in the shallow inshore waters (5 - 10 cm depth), and if possible in deeper water and offshore. A higher temperature inshore than offshore may be a sign of poor water circulation; a lower temperature may indicate groundwater seepage or other freshwater input.

3. Determine the light penetration in the water with a secchi disc (Figure 26, page 56). Lower the disc from a boat or wharf until it is no longer visible from the surface, and note the depth from the marked rope. Slowly pull it up again until it is just visible, note the depth again, and average the two measurements. This light penetration depth is a good measure of turbidity caused by pollution or suspended sediments.

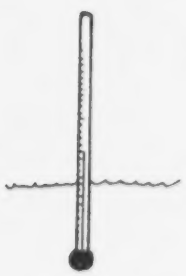
4. Have the students walk along 50 metres of shoreline collecting or counting the number of pieces of plastic or plastic objects observed (bags, bottles, packaging, fishing gear, toys, etc.). Calculate the plastic pollution for each site in terms of the weight of the plastic or the number of plastic objects per metre of shoreline. If plastics are irregularly distributed, it may be necessary to make several measurements and average them.

5. Make a similar count of tar balls in the drift along the shore. On a sandy beach, collect the surface sand and tar balls from a 1 metre wide strip of beach perpendicular to the shore. Wash the sample through a sieve or screen with 2mm mesh, sort out the tar balls by hand, and weigh them or count them and measure their average size (diameter). Take 3 such samples at random on the beach. Calculate the weight (or number) of tar balls per metre of shoreline. Tar balls are a good mesure of nearshore or offshore oil pollution.

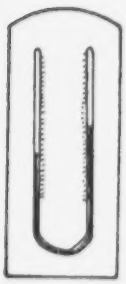
6. If the appropriate equipment is available (Figure 22, page 55), determine the salinity of the coastal water at each site, and compare it with the salinity of seawater from further offshore. If the salinity is lower than seawater, look for signs of freshwater streams or outfalls. If there is no evident sign of freshwater input, there may be groundwater seepage. Such freshwater inputs are important, as they often carry nutrients or pollution from inhabited areas inland.

FIGURE 41

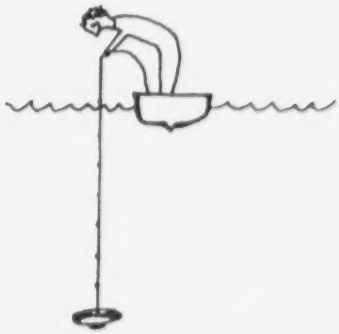
INSTRUMENTS FOR WATER QUALITY ANALYSIS



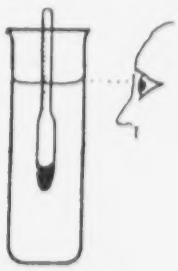
(a) Thermometer



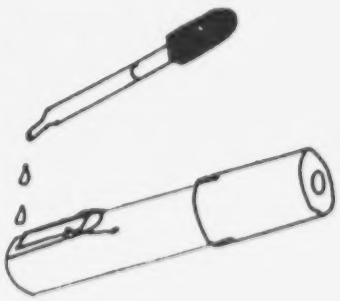
(b) Maximum/minimum thermometer



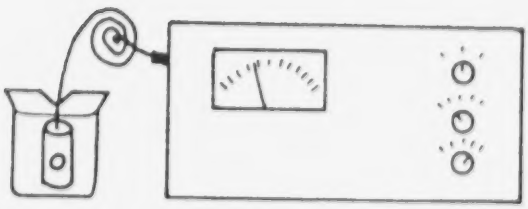
(c) Secchi disc to measure turbidity



(d) Hydrometer



(e) Refractometer



(f) Conductivity meter

7. Prepare a map of the coastline, or other graphic display, and record on it the water quality measurements at each survey point (temperature, turbidity, salinity, plastics and tar balls). Do the signs of water pollution correlate with known activities on the adjacent land?

Teaching hints

If water pollution measurements are available, they can be correlated with the class measurements to make a more complete survey.

Follow up

It can be interesting to compare these water quality measurements with biological measurements along the same shoreline. Activities 1, 2, 3, 5, 7, 12, 13, 17, 18, 19, or 20 would all be appropriate in certain cases, depending on the environment and the effects being studied, to demonstrate the biological effects of a pollution gradient.

This technique can be used by classes each year for long-term monitoring (Activity 32). If signs of possible water pollution are identified, the class may want to present its result to local environment or health officials for possible confirmation and follow-up.

References

SPC/UNEP Coral Reef Monitoring Handbook. United Nations Environment Programme, Reference Methods for Marine Pollution. Studies No. 25, 1984. 25 pp.

UNEP/IOC/IAEA Monitoring of tar on marine beaches. United Nations Environment Programme, Reference Methods in Marine Pollution. Studies No. 15 (draft), 1985. 13 pp.

(Both available from OCA/PAC, UNEP, P.O. Box 30552, Nairobi, Kenya.)

N. G. Willoughby. Man-made flotsam on the strand-lines of the Thousand Islands (Kepulauan Seribu) Jakarta, Java. In Human Induced Damage to Coral Reefs, Unesco Reports in Marine Science. No. 40, 1986. pages 157 - 163.

26. TEMPERATURE TOLERANCE

(A, D; b, c; III)

Objectives

1. to observe the effects of temperature on a marine organism.
2. to determine the temperature tolerance of an organism experimentally.
3. to learn to interpret experimental results in terms of local environmental conditions.

Curriculum links

Temperature, environmental limits, thermal pollution, aquaculture.

Preparation

Habitat: any coastal marine habitat where suitable experimental organisms can be found.

Time required: time for collection of organisms in the field; 2 - 3 hours of laboratory observations; repeat observations after 24 hours.

Teacher: select one or more appropriate common organisms for the experiment. These can be plants or animals, preferably of local ecological or economic significance. If possible the moment of death should be readily observable. Organisms important locally in aquaculture, such as small fish or shrimp, would be ideal. However many marine organisms will not survive in school aquaria, so some preliminary testing will be necessary to find those that will live a week in the laboratory. Intertidal or tide-pool forms may prove most adaptable. Test the experimental set-up to ensure that the organism survives wel in the experimental jars or aquaria before experimenting with temperature variations.

Students: an introduction to temperature effects and temperature tolerances would be useful.

Equipment: dipnet, knife, scraper, shovel for collecting experimental organisms, and bucket for transport
15 aquaria, beakers or glass jars of about 1 litre capacity
supply of clean seawater
thermometers
maximum-minimum thermometer (optional)
5 large basins or water baths in which 3 aquaria or jars can be placed
ice
source of heat or hot water (or electric aquarium heaters)

Strategy

Question: What is the temperature tolerance of a marine organism and how can it be determined?

1. Every organism has temperatures above and below which it cannot live. This temperature tolerance determines the habitats in which it occurs. Organisms in the intertidal are often exposed to greater temperature extremes than subtidal organisms. Tropical organisms may be less tolerant than temperate organisms, and may be living close to their upper temperature limit.

2. Collect the experimental organism(s) selected and measure the temperature in their natural environment. If possible determine the range of temperatures to which they are normally subjected by looking at temperature records or by leaving a maximum-minimum thermometer in the environment for some time.

3. Transport the organisms rapidly to the laboratory, and hold them for 1 or 2 days to allow them to acclimate to the aquarium conditions.

4. Set up a series of water baths to hold the jars or aquaria. Ideally there should be at least 5 water baths, each able to hold 2 or 3 small jars or aquaria containing several organisms (10 organisms per temperature would be good minimum if possible). One water bath (the control) should be the ambient temperature which should correspond to the temperature in nature. The other water baths should be heated (with hot water) or cooled (with ice) to 5°C and 10°C above and below the ambient temperature. The students should follow these temperatures closely and adjust them as necessary during the experiment. Make graphs of the temperatures to which the organisms are subjected.

5. At the start of the experiment, all the jars should contain organisms at ambient temperature. Place the jars in the water baths, and follow and record the temperature in each jar carefully. Once the temperature in the jar reaches the experimental level (of the water bath) it should be kept there for a half an hour, and then removed back to the ambient temperature. Gentle stirring may help the temperatures to equilibrate more quickly.

6. Notes should be made on the state and behaviour of all the organisms during and after the experiment, as related to the temperatures experienced. Check the state of the organisms again 24 hours later for signs of delayed mortality, before ending the experiment. Note the number of organisms surviving in each experimental condition and in the control. What do the results say about the temperature tolerance of the organism relative to its occurrence in nature? If it is an aquaculture organism, are the results useful in planning aquaculture facilities?

7. If it is possible to have enough experimental organisms and replicate conditions, the results can be tested for statistical significance, or even analysed on a microcomputer.

8. The experiment can be repeated with different organisms to compare temperature tolerances. For instance organisms from tide pools or shallow water might be expected to have higher= temperature tolerances than deeper water forms.

27. POLLUTION EFFECTS

(A; c; III)

Objectives

1. to observe the effects of polluted water on an organism.
2. to learn the principles of toxicity testing.
3. to relate laboratory experiments to field observations.

Curriculum links

Pollution, toxicity, environmental impact.

Preparation

Habitat: any coastal marine habitat where pollution effects are apparent in some areas.

Time required: 2 - 3 hours of field observations and two 2-hour laboratory exercises.

Teacher: select an appropriate experimental marine organism that is common, easy to maintain in laboratory aquaria for a few days, and if possible is absent in known polluted areas but present in environmentally-environmentally-similar unpolluted areas. It can be the same organism as that used for the temperature tolerance test. The moment of death should be readily observable. Identify two coastal areas which are environmentally similar, except that one is polluted and the other is not. The outfall or source of pollution should be evident and accessible if possible.

Students: an introduction to the concept of pollution and pollution effects is necessary.

Equipment: dipnet, knife, scraper, shovel for collecting experimental organisms
field notebooks and pencils
15 aquaria, beakers or glass jars of about 1 litre capacity
supply of clean seawater

Large bottles or buckets for polluted water
graduated cylinderor other volumetric
measure of 1 litre
labels for aquaria
thermometer

Strategy

Question: What is the effect of pollution on a marine organism?

1. Pollution is by definition harmful to at least some organisms. Many kinds of water pollution are toxic and kill more and more organisms as their concentration increases. Polluted waters tend to have fewer kinds of organisms than unpolluted waters. While most techniques for measuring pollutants are too complex for secondary school use, it should be possible to observe the direct effect of polluted water on sensitive organisms.

2. Visit the unpolluted intertidalfield site and observe and record the organisms and communities present. Collect about 50 experimental organisms and allow them to acclimatise to the aquaria in the laboratory.

3. Then visit the polluted site and compare the biological communities with the unpolluted area. Collect a large sample (10 litres) of the polluted water from at or near the source. If the pollution source is of low salinity (freshwater), collect the sample from far enough away that some mixing has occurred, to avoid confusing salinity effects and pollution effects in the experiment.

4. In the laboratory, set up 5 sets of aquaria with three replicates per set. One set should have undiluted polluted water, the next diluted by half with pure seawater to 50%, the third diluted by half again to 25%, and the fourth again diluted 12.5%. The last should have pure seawater as a control. check that the temperature is the same in all aquaria, and that this is close to the temperature in nature.

5. Place some experimental organisms in each aquarium, and observe the immediate effects on behaviour and survival.

6. Check the aquaria again after 24 hours and record the number of surviving test organisms as compared with the controls.

7. Discuss what the results demonstrate about the toxicity of the pollutant. Relate the results to the observed distribution of organisms in the field.

Teaching hints

The experimental details, including the dilutions used, many need to be adjusted to the organism used and the type and concentration of pollutant. Some preliminary trials may be needed to select the organism that best demonstrates the pollution effect. Many pollutants are only released intermittently, so the sample should be taken when the pollutant is known to be present.

In places with little local pollution, this activity can still be done using an artificial pollutant such as copper sulphate.

Follow up

The effect of the pollutant on the environment can be demonstrated more clearly with Activity 33, comparing the poiluted and unpolluted areas for environmental impact.

28. FOOD FROM THE SEA

(A; a; I, II)

Objectives

1. to observe what kinds of food are taken from the sea.
2. to measure how much food is taken from the sea.
3. to learn something about fishing techniques and the transport of marketing of seafoods.
4. to understand the importance of the sea to local diets.

Curriculum links

Exploitation of natural resources, fishing.

Preparation

Habitat: any coastal marine habitat where fishing takes place, including if possible a fishing port and fish market.

Time required: one or more visits to observe fishermen's landings and themarketing of fish.

Teacher: obtain any existing statistics on local fish catches. If possible obtain the co-operation of local fishermen in explaining their fishing techniques and showing their catch.

Students: review general information on fishing boats, fishing gear and fishing ports. The principles of harvesting a wild resource, and such measures as catch per unit effort, should be taught together with this activity.

Equipment:
field notebooks and pencils
balances or scales for weighing fish
plastic bags or buckets
hand lens or dissecting microscope (optional)
knife
forceps

Strategy

Question: How important is food from the sea locally?

1. Many coastal communities depend on the resources of the sea to provide an important part of the local diet. This activity allows students to measure that importance for themselves.

2. Visit the local landing place as the fishermen come in with their catches, and/or the local fish market. Observe the fishermen separate the marine animals into various groups: economic fish, trash fish, prawns, crabs, etc. Discuss with the fishermen what kind of habitats they fished in and what gear they used.

3. List the names or descriptions of all the different kinds of seafood brought in or available for sale. If some fisheries are seasonal, try to complete the class data from other sources. If possible take examples of the different animals and seaweeds back to the classroom for further examination and identification. (If the collections cannot be worked on immediately, they can be preserved in 10% formaldehyde, and then washed before student use.)

4. Each student should also make a list of the seafood eaten at home over a period of a week. If subsistence fisheries or gleaning by women and children are important, these also should be observed and recorded.

5. If weighing devices are available (the class can make simple balances), quantify the amount of seafood taken in a given time (say 1 week) by weighing fishermen's catches and determining where they came from and how long it took to catch them. Home consumption of seafood can also be weighed over a week.

6. On the basis of the figures collected, make simple estimates of the amount of food produced locally from the sea, perhaps measured per kilometre of coastline, on an annual basis. If the data are available, also determine the fishermen's catch per unit of effort (hours spent fishing per fisherman or per boat).

7. Compare the local catch or consumption of fresh seafood with that of imported seafood (canned, dried or frozen) using import statistics or students' reports of home consumption. Has the proportion of local and imported seafood eaten changed over the years?

8. If there has been a change in seafood consumption, is it due to a change in the local resources (i.e. from over-fishing) or to consumer preferences (better taste, or the ease of opening a can).

9. Discuss the results of the class study in terms of the management of the coastal marine environment.

29. SPAWNING OF BIVALVES*

(D; b; III)

Objectives

1. to learn techniques for inducing spawning in bivalve molluscs.
2. to observe the behaviour of bivalves releasing sperm and eggs.
3. to understand some methods of bivalve aquaculture.

Curriculum links

Reproduction, aquaculture.

Preparation

Habitat: any coastal area where bivalves are present, especially where they are used for aquaculture.

Time required: field trip to collect bivalves, plus classroom exercise for up to 6 - 10 hours or more, depending on the species.

Teacher: Identify common molluscs, especially bivalves accessible on the shore or those used locally for aquaculture, which are appropriate for this activity. Determine the spawning season of the bivalves to select the time when they will be mature and ready to spawn.

---

*Based on an activity devised by Dr. Twee Hormchong.

Students: Review concepts of bivalve reproduction, including the anatomy, growth and sexual development of the seed bivalves, and processes of fertilisation and larval development. Understand the economic importance of bivalves and their usefulness in aquaculture.

Equipment:

small aquaria or big round glass jars
heater
thermometers
ice
small plastic bags
rubber bands
selected bivalves
compound miscroscope
droppers
microscope slides and cover slips

Strategy

Question: How are bivalves reproduced in large numbers for aquaculture?

1. Bivalves such as oysters, clams and mussels are some of the most important economic marine animals in coastal areas and are widely grown in aquaculture. They are filter feeders that effectively convert plankton into useful animal protein with a high value for domestic consumption and export. Getting them to reproduce at will is an important first step in producing large numbers for aquaculture, followed by raising the larvae until their settlement as young shellfish. This activity shows how to get bivalves to release their eggs and sperm for fertilisation in the seawater.

2. Clean the aquarium or round glass jar. Fill it with seawater which has been sterilised by heating to 50°C for 24 hours or by boiling for 5 minutes, and then cooled to room temperature.

3. Select 4 to 6 bivalves at reproductive maturity, set them in the aquarium or other container, and fill it about $\frac{2}{3}$ full with treated seawater.

4. After 2 hours, put some ice in a small plastic bag, tie it with a rubber band, and put it in the aquarium until the temperature drops to 16 - 18°C. Keep the water at this temperature for 2 hours by adding and removing the ice bag.

5. After 2 hours, add hot seawater little by little to increase the temperature up to 30 - 32°C, and keep this temperature constant for 2 hours.

6. Again add the ice bag to lower the temperature to 16 - 18°C. Continue to alternate between 16 - 18°C and 30 - 32°C at 2 hour intervals until the bivalves release filament-like strands of sperm and eggs.

7. Collect sperm and eggs with an eye dropperm mount them on a microscope. Is it possible to observe fertilisation?

8. Discuss the factors that affect reproduction in molluscs. How long does it take for the fertilised eggs to become larvae?

Teaching hints

Alternative methods for inducing spawning in bivalves are:

- alternating low and high salinity;
- injecting chemicals like hydrogen peroxide (H_2O_2) etc.

30. OVERFISHING

(C; a: I, IV)

Objectives

1. to observe the characteristics of an overfished area.
2. to relate human pressures to their impact on a natural resource.
3. to consider possibilities for the restoration and management of an overused resource.

Curriculum links

Resource use and misuse, fishing, resource management.

Prepaation

Habitat: any coastal marine habitat where overfishing is a problem.

Time required: several 2 - 3 hour periods, depending on methods used.

Teacher: identify a coastal area where overfishing is known to be a problem and fish catches have decreased, and if possible an environmentally similar area where fish catches are normal. Select the activities appropriate to the area that will best demonstrate the effects of overfishing.

Students: prior discussion of the concepts of fisheries management would be useful.

Equipment: see activities referred to in Strategy.

Strategy

Question: What are the effects of overfishing on the environment and how can they be corrected?

1. Overfishing which reduces the productivity of local fisheries is a common problem in many places. Several activities in this manual can be used together to demonstrate the problem and suggest possible solutions. The study can be done just in the overfished area, or preferably by comparing an overfished and an undisturbed or normallyfished area.

2. Using Activity 28 (Food from the Sea), quantify the problem of overfishing, both in terms of catch per kilometre of coastline and of catch per unit of fishing effort.

3. Activity 20 (Reef Fish Survey) can be adapted to the coastal habitat and the most commonly fished species, and used to measure the existing fish populations.

4. One of the activities on zonation and community structure (Activities 2, 3, 4, 5, 7 and 9) can be used to characterise the fishing habitat and possibly to identify environmental changes related to overfishing.

5. Activity 11 (Trophic Levels) can be used to identify important links in the food chain supporting the species fished. Overfishing may affect the balance and structure of caostal food chains.

6. If declining catches may also be related to other factors, a water quality survey (Activity 25) or study of pollution effects (Activity 27) may be appropriate.

7. The overfished area may be compared with an undisturbed area, such as a marine reserve or remote and less frequently visited site, using the environmental impact approach (Activity 33).

8. Discuss the significance of overfishing to local subsistence and the economy, as well as to the productivity of coastal marine ecosystems.

9. On the basis of the data collected fromthe different activities, prepare a list of recommendations to control overfishing and to improve the management of the fisheries resources in the area.

Follow up

The results of the class study and of any recommendations made can be communicated to local fishermen and to fisheries department officials.

An overfished area can also be monitored (Activity 32) to observe changes taking place as a result of fishing or of management efforts to correct the problem.

If the local fishermen agree, a marine reserve (Activity 35) could also be created in part of the area.

31. TRADITIONAL KNOWLEDGE

(A, D; a; IV)

Objectives

1. to collect traditional knowledge about marine resources.
2. to appreciate the value of traditional environmental management.
3. to see how traditional knowledge can help to solve modern problems.

Curiculum links

Environmental management, fishing, traditional cultures.

Preparation

Habitat:	any area where traditional peoples have long-established customs and fishing practices.
Time required:	2 periods of class discussion plus research by students in their own families or villages.
Teacher:	collect whatever information is available on local traditional knowledge of marine resources and practices, such as anthropological or ethnographic studies.
Students:	a review of different methods of fisheries management would be useful background in recognising the value of traditional approaches.
Equipment:	field notebooks cassette recorders for recording interviews (optional) camera for photographing fishing gear and other artifacts (optional)

Strategy

Question: How did the traditional culture use and manage marine resources, and of what value is that knowledge today?

1. The original inhabitants of most coastal areas lived close to the sea and depended on it for survival. They observed nature closely, and over generations came to understand many things about the natural world and how it worked. Most villages had their wise old men and women to whom others would turn for advice and who would decide when and where to fish. Many traditional practices were developed where necessary to ensure adequate food for the people and to protect resources from overuse. In general these systems worked well until the arrival of the Europeans brought new techniques and new economic pressures. The old knowledge and values were often discredited as sorcery or superstition, and children went off to school instead of learning from their elders. Much of this practical knowledge has been lost as old people have died without passing it on to the next generation. Often only fragments are still remembered, or have been recorded by outside observers. Yet much of this information would be useful to the wise management of resources today. It is important to try to save what is left of this traditional knowledge and the value system of respect for nature on which it was often based, so that it can be reinterpreted in the light of modern scientific understanding, and reapplied as necessary to manage resources better.

2. Discuss with the class the significance of local traditional management practices in the marine environment. This could include such things as:

   Fishing methods and materials.
   Knowledge of fish species, their local names, behaviour, migration and reproduction.
   Best fishing locations, times and techniques for each species.
   Controls on fishing: limited access to fishing areas, prohibited (taboo) areas or seasons, catch restrictions, permitted fishing methods.
   Changes in fishing resources, effects of over-fishing, "how things used to be".

3. Students should try to collect as much traditional knowledge of the environment as possible, either locally in their families, or over vacation periods if they return to their home areas or villages. They should ask the elders to recount oral traditions, and to demonstrate traditional ways or making fishing gear. If possible, they should participate in fishing expeditions or other trips where they can learn traditional knowledge first hand.

4. The information collected should be recorded as written notes, or as cassette recordings or photographs if the means to do so are available. If such knowledge is traditionally kept secret within the family, the details need not be shared with the class. What is important is that it is transmitted and appreciated.

5. Another class discussion should be held to report on the results of the student papers or projects.

6. The class can prepare recommendations as to ways in which traditional methods or experience may help to improve the modern management of fisheries resources.

Follow up

The information collected could well be of interest to the local museum or cultural centre. If so, the class can prepare carefully documented archives for deposit in the appropriate institution. The recommendations can also be presented to the elders who co-operated in the projects and to local fishermen.

References

Modified from Training Materials for Rural Enviromental Management prepared by A. L. Dahl for the South Pacific Regional Environment Programme (1984).

For specific examples, see:

Johannes, Robert E. Traditional marine conservation methods in Oceania and their demise. Annual Review of Ecology and Systematics (Palo Alto), Volume 9, 1978, pages 349 -364.

Dahl, Arthur Lyon. Traditional environmental management in New Caledonia: a review of existing knowledge. South Pacific Regional Environment Programme Topic Review (Noumea), No. 18, 1985, 17p.

32. MONITORING

(A, C; a, c, d; II, IV)

Objectives

1. to study stability or change in an environment over time.
2. to observe effects of seasons, weather or population cycles.
3. to learn how to follow the environmental impact of a development project over time.

Curriculum links

Environmental stability and change, seasonality, environmental impact assessemnt.

Preparation

Habitat: any coastal habitat where change may be taking place.

Time required: depends on frequency of monitoring and activities used.

Teacher: choose what is to be monitored locally: seasonality; long-term changes from year to year; the effects of a biological phenomenon such as a population explosion (e.g. Acanthaster Crown-of-thorns starfish) or die-off (e.g. Diadema sea urchins); recovery of the environment after storm damage; or the effects over time of a development project or other environmental change. The technique to be used and its frequency will depend on the type of change and the speed with which it is expected to take place. For rapid short-term changes, the class can make 2 or 3 monitoring surveys during the school year. However most environmental change is gradual, so the activity should be set up with permanent plots where each class survey can be compared with the results of classes in previous years.

Students: the value of repeating a survey to detect changes should be understood. Care must be taken to do exactly the same thing each time the survey is made.

Equipment: same as for the survey technique chosen.

Strategy

Question: What is changing in the environment, if anything?

1. Several activities in this manual describe survey methods that can be used for environmental monitoring. It is simply necessary to repeat the same survey at the same location at regular intervals and to compare the results to see if there has been a change in the environment.

2. The following activities will provide information for monitoring:

   2. Zonation in a sandy-muddy area
   3. The mangrove community
   5. Rocky shore zonation
   6. Zonation on mangrove trunks
   7. Area coverage on a coral reef
   12. Seagrass turnover
   20. Reef fish survey
   25. Water quality survey

   Select the activity and technique that corresponds best to the problem to be monitored. Quantitative techniques will give better results for monitoring than simple qualitative techniques. Greater care in the survey will permit more confidence in the results.

3. Some examples of monitoring activities would be:

   (a) an annual survey to see if long-term changes are taking place in the environment;

   (b) surveys every 4 months around a coastal construction project to see if it is changing the environment;

   (c) surveys every 3 months to see if there are seasonal changes in certain populations;

   (d) surveys every 2 months to observe recovery after severe storm damage;

   (e) surveys each year of a reef affected by Acanthaster to record any decline or recovery of living corals.

4. The area to be monitored should be permanently marked, preferably with several firmly-fixed steel rods or stakes at precisely measured locations, so that each survey is made in exactly the same place. Otherwise sampling error will make it difficult to detect real changes. If one marker is lost, it can usually be replaced by reference to the other markers.

5. Some monitoring activities can be completed with a school year, and the class can see and interpret the results of their efforts. Others may require more than one year to produce results. ach class (except the first) can then compare its results with those obtained in prior years. It is important for the school to keep a permanent record of the results, of the methods used, and of the places studied.

6. The results of the monitoring surveys should be discussed in class, and any changes observed should be related to their possible causes. Some variability may be due to sampling error, some to natural changes, and some to the effects of human activities.

7. If sampling error is suspected, test this by having the class repeat those parts of the survey in the same place at short intervals without looking at the prior results, to see how much variation is due to the survey method.

Teaching hints

Interpreting the results of a monitoring survey is never easy, as many causal factors may be involved. This uncertainty itself should be discussed in class.

A government officer or scientist might be interested in co-operating with the class monitoring survey, and could perhaps explain the results to the class and show their importance for management.

Follow up

Well-documented monitoring surveys can produce results of great interest to fishmen, scientists and government departments responsible for the management of coastal areas and fisheries. The class could prepare a report for submission to the community or to the appropriate authorities.

33. ENVIRONMENTAL IMPACT

(C; a, b, c, d; IV)

Objectives

1. to observe the differences between a disturbed and undisturbed environment.
2. to evaluate the effects of development on the environment.
3. to understand the process of environmental impact assessment.

Curriculum links

Environmental impact assessment, development and environment.

Preparation

Habitat: any coastal marine habitat where impacts of development can be compared with a similar undeveloped or undisturbed area.

Time required: 2 - 3 hours of field work at each of two sites, depending on the methods chosen.

Teacher select two sites which are (or were) as similar as possible in terms of environmental conditions and biological communities, but in which one has suffered the obvious effects of a development project or other impact, and the other is still undisturbed. Decide which activities will best illustrate the nature and extent of the impact.

Students: discuss the different kinds of effects of human activities or development on the environment. The procedures used nationally or internationally for environmental impact assessment can also be reviewed.

Equipment: as required for the activities chosen.

Strategy

Question: What are the environmental effects of development projects and how can they be measured?

1. The assessment of the environmental impact of development projects in order to anticipate and avoid harmful environmental effects is now widely accepted as a standard part of the planning process. Impacts can be measured by studies before and after the development, or by comparing impacted and unimpacted areas. Potential impacts are projected on the basis of the observed effects of similar projects or kinds of disturbance elsewhere. The comparison of impacted and unimpacted areas will allow students both to observe the effects and to experience the process by which impacts are measured or anticipated.

2. Depending on the effects to be observed, choose one or more of the following activities:

    1. Collections of marine life
    2. Zonation in a sandy-muddy area
    3. The mangrove community
    5. Rocky shore zonation
    6. Zonation on mangrove trunks
    7. Area coverage on a coral reef
    9. Coral reef surface area
    11. Trophic levels
    12. Seagrass turnover
    13. Mangrove leaf production
    17. Coral growth rates
    18. Animal population sizes

19. Settlement and succession
20. Reef fish survey
25. Water quality survey
27. Pollution effects
28. Food from the sea.

3. Carry out the same activities both in the impacted area and in the undisturbed area, and compare the results.

4. Discuss and interpret the results in terms of the effects on species and ecosystems of the types of impact demonstrated to have occured in the area.

Follow up

An environmental impact study can be followed by monitoring (Activity 32) to observe if the impact continues or changes over time, or if there is gradual recovery.

34. CONSERVATION SURVEY

(A, D; IV)

Objectives

1. to learn how to make a survey of resources.
2. to determine the priority areas of coastline for conservation action.
3. to understand the importance of conservation as part of development planning.

Curriculum links

Conservation, coastal zone management, planning.

Preparation

Habitat: all the coastline within reach of school field activities.

Time required: 2 - 3 hours of field work per site surveyed; at least 5 sites should be studied, more if the coastline is long or includes many different habitats

Teacher: this activity can integrate information from other activities carried out during the school year, although some additional surveys will almost certainly be required. Class field work can be planned over the whole year with

a conservation survey as a final result. The survey can also be made as a separate exercise. Prior to the field work, obtain a good map of the coast, preferably with details of coastal habitats like mangroves and coral reefs. If aerial photographs are available they willalso be very useful.

Students: basic concepts of conservation of nature, and of coastal zone planning and management, should be understood.

Equipment: maps and/or aerial photographs of the coastline
large sheets of tracing paper
coloured pens or pencils
field equipment as required by activities selected

Strategy

Question: What areas or ecosystems along the coast have the greatest conservation interest?

1. No country should allow development to destroy all of its natural resources, habitats and ecosystems, which are very important for the future. It is essential that viable examples of each kind of natural system should be conserved,usually in some type of protected area, park or reserve. Coastal ecosystems therefore need to be surveyed for their conservation interest, so that the best or most important examples can be protected for the future. This activity involves identifying the most interesting coastal areas near the school, as a contribution to conservation planning.

2. On the basis of maps, aerial photographs, and personal knowledge of the coast, select a series of survey sites representative of the different types of coastal habitats and features, of spaced at regular intervals along the coast (every 1 to 5 kilometres, depending on the regularity of the coastline). Include any sites known to be of particular interest or biological richness, to have rare or endemic species, or to be critical habitats where organisms come to reproduce.

3. Using the approach described in Activity 1 (Collections of Marine Life) minus the individual collections, prepare a species list for each survey site. Rank the sites according to the number of species found. If any species are of particular rarity or conservation interest, note this separately.

4. Collect information on the ecological interest and richness of each site using those of the following activities that are appropriate to the type of habitat:

   2. Sandy-muddy shore zonation
   3. The mangrove community
   4. Coral reef structure
   5. Rocky shore zonation
   7. Area coverage on a coral reef
  18. Animal population sizes
  20. Reef fish survey

   For each type of habitat (coral reef, mangrove, rocky or sandy shore, lagoon, etc.), determine which are the richest and most interesting ecologically, and rank them in order.

5. Sites with a greater diversity of habitat types should generally rank higher than those with only one type of habitat.

6. If some parts of the coast have been damaged by pollution or development, a water quality survey (Activity 25) at each site will help to show which are the most natural and least disturbed. If old maps or aerial photographs are available, they can be compared with recent information to show what natural areas have already been damaged or lost, particularly to dredging, filling or other coastal construction.

7. Using the different kinds of information collected, the class should select those areas with the greatest interest and importance for conservation, that best represent the coastal resources of their country or region.

8. A large map of the coastline can be prepared showing the distribution of the different coastal habitats, the location of the survey sites, the species numbers, habitat rankings, water quality and other information collected. Areas with the greatest potential for creating marine protected areas can be highlighted.

9. Discuss with the class the importance of a conservation survey as part of comprehensive planning for the management of the coastal zone.

Follow up

Copies of the class conservation survey and map can be presented to local leaders and appropriate govenment officials.

35. A MARINE RESERVE

(C; d; III, IV)

Objectives

1. to create a marine reserve.
2. to observe the effects of protection on species and ecosystems.
3. to appreciate the values of protected areas for resource management.

Curriculum links

Conservation, coastal zone management, fisheries.

Preparation

Habitat: any coastal marine habitat that is already protected or that can be protected as a school reserve.

Time required: 2 - 3 hours of field observations each 1 - 2 months.

Teacher: if a marine protected area already exists near the school, obtain permission from the responsible authorities to study some part of the area. If there is no such area, obtain the approval of local authorities and/or fishermen for the creation of a small school reserve in an accessible coastal area.

Students: background information on conservation and the role of protected areas.

Equipment: signs and markers to designate the protected area field equipment as required for activities selected.

Strategy

Question: What is the value of a protected area?

1. Protected areas such as marine parks and reserves are an important pat of national conservation and development plans. They preserve undisturbed examples of natural ecosystems, and may serve as a refuge and breeding area where species that are fished or collected can multiply and repopulate adjacent exploited areas. If there is no existing marine protected area, select an appropriate site as determined by accessibility to the school and/or conservation interest as determined by a conservation survey (Activity 34). An area approximately 100 - 300 metres on a side should be sufficient for the school project, although a larger area would be of greater value for conservation.

2. Obtain the agreement of local and/or national authorities and local fishermen and land owners that the area will be considered as a school nature reserve, and that fishing and other exploitative activities will not be done in the area, at least for the school year, if not for an indefinite period. The site could be legally set aside as a protected area, but protection may be equally effective if it is by informal agreement with local fishermen and other users.

3. Mark the boundaries of the area with signs and marker poles so that people are reminded of its protected status.

4. Survey the school reserve using the appropriate activities, but avoiding any collections or damaging activities. Survey activities may include:

   2. Sandy-muddy shore zonation
   3. The mangrove community
   4. Coral reef structure
   5. Rocky shore zonation
   7. Area coverage on a coral reef
   18. Animal population size
   20. Reef fish survey.

5. Repeat the survey every 2 - 3 months to observe the effects of protection on the species and communities. Note particularly the status of any species that were previously fished or exploited.

6. If there is a similar but unprotected area not too far away, make comparative surveys to show the differences between the protected and unprotected areas.

7. Surveys of fish (Activity 20) and food from the sea (Activity 28) can be made or repeated in adjacent waters to see if the existence of the reserve had led to better fish populations and catches outside the reserve.

8. Other marine activities can also be carried out in the school reserve as long as they respect its reserve status and do not involve collecting or damaging organisms.

9. Prepare a class report on the reserve and on the effects of its protected status, to be shared with local fishermen and authorities.

Follow up

If the class studies have demonstrated the effectiveness of the school reserve, try to have it established on a permanent basis.

36. MARINE AQUARIUM

(B, D; b, d; III)

Objectives

1. to learn methods of rearing marine fishes in an aquarium.
2. to observe fish behaviour.
3. to understand the complexity of maintaining a suitable environment for marine life.
4. to appreciate the value and beauty of marine aquaria.

Curriculum links

Animal behaviour, effects of physical and biological factors, nutrition, mortality.

Preparation

Habitat: fish may be collected from any coastal environment.

Time required: 2 - 3 hours to set up aquarium, one or more field trips to collect fish (optional), plus regular daily care.

Teacher: obtain and study reference works on rearing marine fishes in an aquarium (often available from pet or hobby shops). Determine the best supply of marine aquarium fishes: commercial purchase, local collectors, or collection by the class at a suitable site. Check on any government regulations concerning aquarium fish collecting. If there is a marine aquarium in the area, arrange for a class visit.

Students: review the principles of marine fish rearing in aquaria. Organise in groups of 4 - 5 to take on the different responsibilities for the aquarium.

Equipment:
inexpensive aquarium of glass, plastic or plexiglass with walls 13 - 19 mm ($\frac{1}{2}$ - $\frac{3}{4}$ in) thick
sea water of 30 ppt salinity and pH 7.9 to 8.5
water filter set (if available)
inexpensive air pump
light (ordinary electric or fluorescent lamp)
dip net
marine aquarium fishes
marine fish food
disenfectant: formaldehyde, copper sulphate, methylene blue, malachite green and/or potassium permanganate
medicines as needed for disease treatment
thermometer
pH test kit or meter
salinity measuring device

Strategy

Question: How can marine fishes be raised in an aquarium?

1. Prepare the aquarium by cleaning it. Purify the sea water by filtering; check that the salinity is about 30 parts per thousand (ppt) and the pH between 7.9 and 8.5. Place a considerable amount of sterilised sand, stone,coral sand or gravel in the bottom of the aquarium. Fill the aquarium about $\frac{2}{3}$ full with treated sea water. Aerate the aquarium with a small air pump. Filter the water constantly to keep it clean, and clean the filter regularly.

2. Obtain mature marine fishes or collect them from a suitable coastal site, disinfect them so that they are free from diseases and parasites, and put them into the prepared aquarium at an average density of 1 - 2 fish per 10 litres (3 - 6 fish per foot^3). For large fish the density should be less.

   Some kinds of marine aquarium fish are:

   - beautiful fishes: butterfly fish, angel fish, parrot fish, red soldier fish, wrasses and batfish;
   - coral fishes or sea anemone fishes: clown fish, pink skunk anemone fish, yellow tailed anemone fish, anemone fish, etc.;
   - strange fishes: trunkfish, pufferfish, triggerfish, spiny puffer, procupine fish, lion fish, eels, rays, sea horses, etc.;
   - economic fishes: giant grouper, grouper, red snapper, sea bass, milk fish, etc.

3. Arrange the lighting according to the kinds of fish and where they are found: clown anemone fish, butterfly fish and angel fish prefer strong light; seahorses, batfish and groupers prefer mediumlight; and giant groupers, eels and lionfish prefer low light.

4. Feed the fish daily with food appropriate to the kinds of fish:

   - fresh food: fish, shrimp and oyster flesh;
   - living food: shrimp larvae, Artemia;
   - planktonic food:
     - phytoplankton: Chlorella, Isochrysis, Chaetoceros;
     - zooplankton: protozoa, rotifers, Artemia;
   - Commerical instant marine fish food.

5. Clean the aquarium after feeding.

6. Keep a close watch for fish diseases and treat them if necessary:

Disease	Treatment
Viral/bacterial diseases	- separate diseased fish - treat with antibiotics
Parasitic diseases:	
- fungus: Saprolegnia	- separate diseased fish - sterilise aquarium with copper sulphate or formaldehyde - treat with antifungal medicine
- dinoflagellate: Oodinium	- same treatment as for fungus
- external protozoa: Costia	- separate diseased fish - disinfect aquarium with formaldehyde, copper sulphate, methylene blue or malachite green
- intradermal protozoa: Ichthyophthirius	- separate diseased fish - sterilise with formaldehyde, malachite green or copper sulphate - raise water termperature as high as fish can endure
- intestinal protozoa: Hexamita	- apply Enheptin
- external trematoda: Gyrodactylus	- immerse in formaldehyde or potassium permangante
- parasitic worms: trematoda, cestodes, nematodes and acanthocephatids	- eradicate hosts or intermediate host such as gastropods or crustaceans and apply Kamala for cestodes and Piperazine citrate for round worms.
- crustanceans: Ergasilus	- pinch with pincers or immerse in potassium permanganate or formaldehyde.

7. Observe and record fish behaviour such as feeding activities, group behaviour, mating and reproduction.

8. Record changes in each fish such as growth, colour, appearance, behaviour, etc.

Teaching hints

Supervise the student activities closely, as one mistake can result in the death of all the fish. Consult a local marine aquarium specialist for details on the preparation of disinfecting solutions and medicines, for the symptoms of the various diseases, and for any other problems that may arise. Check the sea water from time to time for salinity, pH, cleanliness, etc. and take corrective action as necessary.

HUMID FORESTS

INTRODUCTION

Humid forests share with other tropical regions high average temperatures. This combined with heavy rainfall - either all the year round or mainly concentrated in a monsoon period each year - results invariably in a dense forest of high trees, abundant ground flora and profuse canopies. There is no fallow, winter period and growth is continuous. The abundant vegetation provides habitats and resources for a great variety and number of animal species. In all the humid forests contain a greater array of species than any other biome.

Humid forest regions, however, exist in a delicate ecological balance. Their soil is typically red laterite coloured by iron or black earth derived from basaltic rock. It and the water draining the forests are characteristically deficient in nutrients. The forests survive because they have ecological systems whereby nutrients are recycled continuously between soil and plants, within food webs and through decomposition. Much is still to be learned about these systems although we do know, only too well, that exploitation of the forests through over-farming, excessive slash-and-burn methods of shifting agriculture and other forms of mistreatment will quickly turn them into deserts.

The main aim of a practical ecological course is to investigate major ideas such as those outlined in Chapters 2 and 3. However, the special nature of humid forests points to four particular features of which students should be strongly aware.

Species diversity The abundance of life in the forests raises the question: how is it maintained? An awareness of the specific requirements of species and the nature of the ecological niches and communities in which they live is necessary if we are to answer this question.

The importance of the soil community The delicate, often precarious, role that the soil plays in the ecology of a humid forest should be in the forefront of the mind of everyone living in one. Without this awareness the forest can be easily exploited to destruction.

The interaction of living things In regions of the world where species are less numerous a dominant ecological consideration is the relation between living organisms and their physical environment. In humid forests the role of interactions between living things is more often the crucial element in maintaining an ecological balance. The physical environment is, of course, also important but an awareness of the way animals and plants are associated and adapted to living with each other, and the way they depend on one another has particular significance in understanding its ecology.

The human use of the forest Humid forests are rich sources of food, medicines, wood and many other commodities which people use and exploit. As Chapter 3 has shown human exploitation and

management is a key theme in ecology. In humid forests the balance between the two is very delicate. An awareness of this is essential to ensure that people gain optimum benefit from the forests.

Undertaking ecological studies in humid forests can be as rewarding as in any other region, if not more so. However the sheer abundance of life in them can present difficulties. There is so much to sort out. For this reason it is important to be as specific as possible in setting the objectives of field studies and to focus on a few, key ecological principles. Two other aspects should also be borne in mind. The first concerns the identification of plants and animals. With such a variety of species much time can be spent on identification and the students' minds directed away from the ecology they are supposed to be studying. Careful identification is important but it needs to be restricted. Studies should be concerned with a few, easily identified species and simple identification keys (often needing to be prepaed by the teacher) should be used.

The second aspect is the importance of examining and preparing an area before it is used for an activity. If possible use an area close to the school and make sure the students can carry out their work easily.

1. DIVERSITY AND DISTRIBUTION OF VEGETATION IN A FOREST AREA

(A, B; a, d; I, II)

Objectives

1. to describe forest area in terms of species composition, species diversity, tree sizes and stratification.
2. to construct a "slope profile" of the study area.
3. to construct a profile of vegetation distribution in the area.
4. to relate the distribution of vegetation to features of the area such as slope, light, etc.

Curriculum links

Diversity and distribution, ecosystem, ecological balance, plant interactions, conservation, forest management, field techniques.

Preparation

Teacher: Survey of the area and selection of sites (natural rainforest or secondary forest). Secure permits from the head of school and parents. Schedule the trip. Conduct a briefing on what the students should bring, prepare, and be expected to do in the field. Prepare all materials needed for the activity to include medicines for insect bites and first aid.

Students: Review field techniques to be used in activity. Learn how to use a simple identification key for the local flora.

Equipment: (for each group)
A reel of cord/string marked at 1 m. intervals
Stakes
Two 3 m. poles (one marked in 0.5 m. intervals, the other in centimetres)
A spirit level
A clinometer
Tape measure

Strategy

Question: What are the main types of vegetation found in the forest and what effects their distribution?

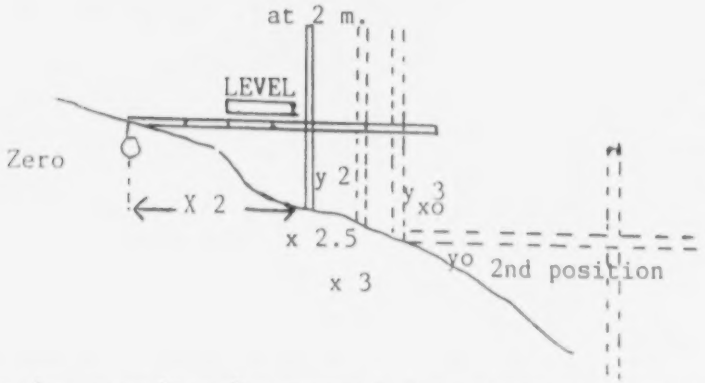
1. Lay out the study area with a string perimeter 60 m long and 10 m wide to produce a belt transect. (See Page 21)

2. Obtain a profile of the slope along a line to one side of the belt transect. Three groups of students can work simultaneously, two at each side of the belt transect and one down the middle.

(a) Start at the higher end of the transect line designated as point O. Lay one pole (the one marked at 0.5 m intervals) horizontally from the O point. Check using the spirit level. At the first 0.5 m mark, stand the other pole vertically. Record the distance of the horizontal pole from the ground. (See Figure 42)

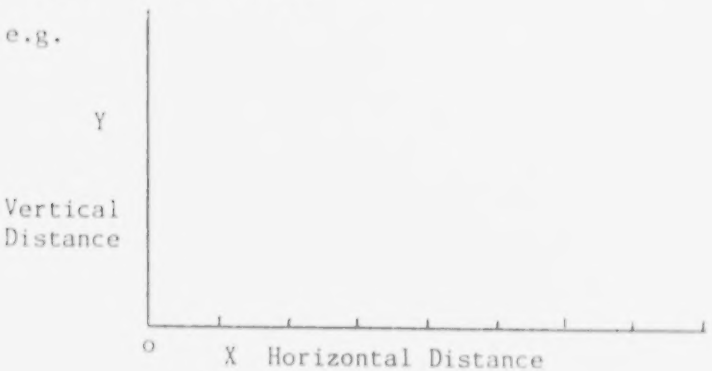
FIGURE 42

OBTAINING A SLOPE PROFILE



Repeat the procedure for every 0.5 m. along the horizontal pole. When the end of the pole is reached, slide the horizontal pole along to the foot of the vertical pole and continue measurement. Make sure the vertical pole does not rest on any stone or object that would give a false reading of the height.

(b) Plot the 2 sets of data on a graph to obtain the profile diagram of the slope.



(c) Moving up (or down) the transect at 1 m. intervals the students should do the following.

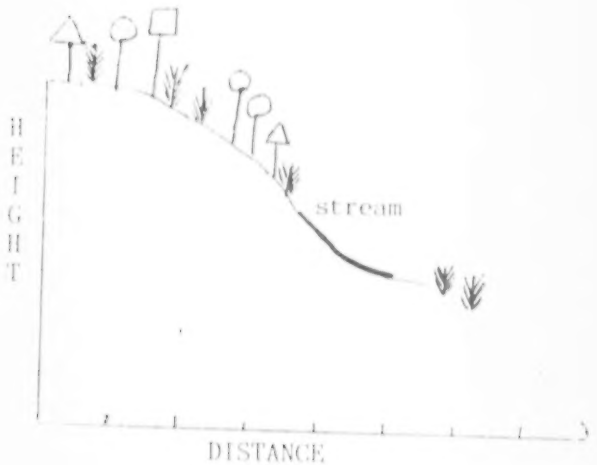
Identify and count each plant species referring to a key or a handbook on the local flora.

For each species measure its height using a tape measure, clinometer, or by estimation based on comparison with plants of known heights.

(d) Construct a profile of vegetation distribution. Assign a symbol for each species. e.g. ↑ conifer; ⩔ - fern; ♀ - palm. All groups must agree on a common symbol for a particular species. Put in the plant symbols in the respective location on the previously made slope profile diagram.

FIGURE 43

EXAMPLE OF PLANT DISTRIBUTION SHOWN ON A PLANT PROFILE



(e) Groups compare results and profile diagrams.
Discuss what are the dominant species in the area?
What is the extent of species diversity? (May include in the activity computation of the diversity index.)
What factors in the environment appear to have determined plant distribution?
Tree sizes and stratification if observed. Identify the canopy species and the subcanopy species and thei role in bringing about the conditions and characteristics of the forest.

Follow up

Further discussion on:

(a) Succession in the forest. Any indications? What would be the forest composition through the successional process?

(b) What if man disturbs or destroys the trees? Predict the effects.

(c) Conservation of forest resources.

2. ECOLOGICAL MODIFICATIONS OF LEAVES

(A, B; I, II)

Objectives

1. to describe diverse forms and characteristics of leaves of plants associated with three different environments; water, moist and dry.
2. to relate the observed structural adaptations to ecological factors especially water and humidity.

Curriculum links

Diversity of form and structure, adaptation of organisms to their environment.

Preparation

Teacher: Survey and select areas for the study. Prepare charts for recording data. If available show technique films on preparation of specimens for microscope work - sections and wet mounts. Demonstrate the separation of epidermal layers of leaves. Provide visuals on the anatomy of the leaf - external and cross-section.

Students: To be aware of three types of habitat especially those closest to the school. To have a knowledge of the names (at least common names) of local plants. To be familiar with the anatomy of the leaf - external and cross-section. To be skilled in the use of the microscope. To be skilled in preparing sections of specimens in wet mount.

Equipment:
small plastic bags
razor blades, sharp knives or a microtome
glass microscope slides
prepared slides of leaf epidermisand leaf cross-section
monocular microscope

Strategy

Question: In what ways is the structure of leaves of plants related to their habitat?

1. Divide class into groups of 4 or 5 students and appoint group leaders. Allot groups to areas of study.

   Area I - an aquatic environment such as a pond, stream, river or lake.

   Area II - a moist, terrestrial environment.

   Area III - a dry, exposed area.

2. In each area, identify the plants that are found, using a local flora (if necessary constructed by the teacher). List them as in Table 4 and indicate their approximate abundance.

TABLE 4

Area	Name of Plant	Abundance (+ few; ++ moderate; +++ abundant)

3. Collect 2 or 3 samples of leaves from each species. Place them in small plastic bags labelled with the name of species and the area from which they were taken. Bring to the laboratory for further observations.

4. In the laboratory, use Table 5 to describe external characteristics of each leaf.

TABLE 5

		Leaf Characteristics				
Area	Name of Plant	Shape (sketch)	Length (mm.)	Width (mm.)	Thickness (mm.)	Other Characteristic

5. Examine prepared slides of leaf epidermis and a cross-section of a leaf under a microscope. Be able to recognise the cuticle, epidermis, stomata, palisade and spongy mesophyll, and support and conductive tissue.

6. Return to the leaf collection. Pick out one species and carefully peel off a piece of the upper and lower epidermis from each leaf. Immediately mount in water on a slide. Observe under the microscope. Count the number of stomata that are visible within the field of view. Move the slide to another spot and count the stomata. Move again until you have counted in 5 fields. Sum up the number of stomata and divide by 5 to get the average number of stomato per field of view. Record the data in Table 6.

TABLE 6

		Number of Stomata					
Name of Plant	Area	1	2	3	4	5	Ave. No. per field

Do the same as above for all the other leaves.

7. Cut very thin cross sections of each leaf using a microtome (if available) or a sharp razor or knife blade. Practice for some time until the thinnest sections possible are obtained. Prepare water mounts of each kind of leaf and observe and record the characteristics listed in Table 7.

TABLE 7

		Leaf Section				
Name of Plant	Area	Cross-sectional shape (sketch)	Thickness of Cuticle	Thickness of epidermis	Presence of palisade layer	Other Observation

8. Discuss and interpret the results. Deduce ways by which the leaves are adapted to their environment.

Follow up

Encourage students to read further in the library on plant adaptations to ecological factors in their environment and to submit a report on it.

3. SOIL AND LITTER COMMUNITY OF THE FOREST

(A, B; a, d; I, II, III, IV)

Objectives

1. determine the categories of soil and litter animals, particularly anthropods, to the level of the genus or family.
2. to determine the trophic relationships of the organisms in the soil and litter layer.
3. to determine the distribution pattern of the major or dominant species.

Curiculum links

Food webs, soil structure, litter decomposition, diversity, animal extraction techniques.

Preparation

Teacher: Select suitable sample sites i.e. those with a well-defined litter layer in the forest. Provide handouts on procedures of activity. Obtain identification keys for invertebrates. Instruct students on the operation of the extraction apparatus. Instruct students on computation techniques required for pattern analysis.

Students: Read handouts given by the teacher. Revise use of identification keys with the help of sample specimens. Revise sampling techniques and computational methods.

Equipment:
Quadrat frame (10 cm^2)
trowel
stakes and raffia
plastic bags
alcohol/ethyl acetate
Tullgren Funnel Extraction Apparatus
Petri dishes
hand lens
microscope

micropipette
fine paint brushes
specimen tubes and wet labels

Strategy

Question: What types of anthropods are found on the forest floor habitat?

1. Locate a transect line on the ground between two stakes and collect litter and soil samples from quadrats aligned along the transect. (Depending upon the size of the class the number of samples collected should ensure that the variety of conditions along the transect are covered.)

2. Using a trowel scoop up the leaf litter and soil up to a depth of 60 mm. from each quadrat. Place the samples in separate polythene bags labelled in sequence.

3. In the laboratory the samples can be treated together or as separate litter and soil samples for extraction in simple Tullgren Funnel extractors in batches. Spread the samples over sieves (about 10 meshes per inch) placed in the funnels. (See Page

4. Heat and light (45 watt bulbs) from above will induce the animals into collecting tubes containing some 70% ethyl alcohol as a perspective.

5. Leave samples in this position for about three days to complete extraction.

6. After extraction pour the contents of each collection tube in turn into a Petri dish for examination and identification of species under a hand lense or microscope. (See Table 8)

7. Identify the organisms up to the family or generic level and record the abundance of each type of organism.

8. Tabulate for each sample of litter and of soil (or combined) the list of organisms encountered and their abundance.

9. Using observations and reference to books categorise the animals into feeding (trophic) groups viz. herbivores, carnivores or omnivores. Then construct a food web linking the individual animals with one another and with, also, the dead plant material (litter) as the starting point.

TABLE 8

DATA SHEET FOR RECORDING DISTRIBUTION OF ORGANISMS

Name of Species	Abundance (Number of individuals) At Each Quadrat Position	Mode of feeding*
	1 2 3	
A	a_1 a_2 a_3	

*state whether herbivorous/detritivorous
carnivorous/detritivorous
omnivorous/detritivorous

Follow up

1. Stimulate discussion on anthropod abundance in the forest soil-litter complex. Are the types of species in the soil different from those in the litter? Why is one species very numerous in one habitat and rare in the other?

2. Is there a tendency for local abundance/aggregation in the distribution of some species? Suggest reasons for this to occur e.g. differences in leaf litter composition, influence of activity of other micro-organisms, presence of suitable air spaces or root exudations.

4. A STUDY OF THE BEHAVIOUR OF EARTHWORMS IN A WORMARIUM

(A, B, D; a, d; I, II, III)

Objectives

1. to describe the behaviour of earthworms in a simulated environment.
2. to use qualitative methods of observation, recording, description and influence.
3. to recognise the importance of earthworms in the soil enriching process.
4. to understand the concept of soil as a dynamic habitat for soil animals.

Curriculum links

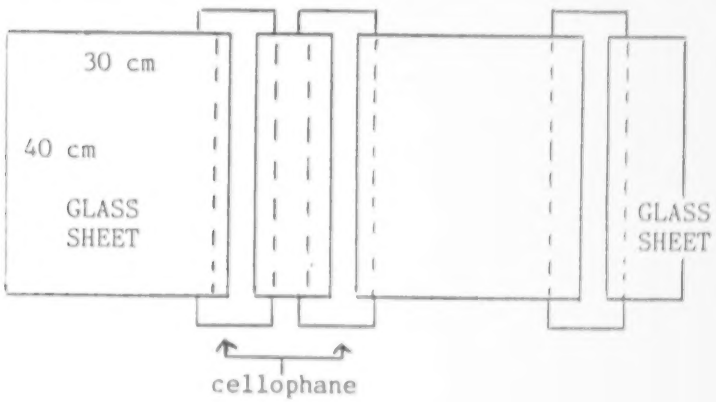
Behaviour, function of organisms in soil, aeration in soil.

Preparation

Teacher: Prepare 2 handouts for students: 'How to construct a wormarium' and 'Observation Schedule'.

How to construct a wormarium

(a) Assemble the glass sheets provided alongside each other and use cellophane to tape them along the edges as shown below.



(b) Fold the glass pieces so that a cuboid is formed and fold back cellophane at the edges. Line bottom with thick plastic sheet and secure with cellophane.

(c) Use the polystyrene piece to make a support base for the wormarium. Stand the wormarium onto the base and secure with plasticene, if necessary. The wormarium is now ready for use.

Observation Schedule

Date	Time	Description of Behaviour	Name of Observer

Select a suitable location for collection of earthworms as well as rich soil. If possible, collection may be made within the school compound itself. Alternatively, students may be instructed to collect soil and earthworms from their own garden. Discuss with students why the collection is best done in the night.

Students: Revise litter and soil structure. Revise earth organisms.

Equipment: For the construction of a wormarium, the following materials are required:

Two piecesof glass sheet of dimension 30 cm. x 40 cm.
Two pieces of glass sheet of dimension 2 cm. x 40 cm.
Cellophane
Thick plastic sheets of dimension 10 cm. x 50 cm.
Polystyrene pieces of dimension 10 cm. x 20 cm. x 50 cm.
Some plasticine (optional)

Strategy

Question: How do earthworms behave in a wormarium?

1. Divide class into groups of 2, 3 or 4 students each.
2. Provide each group with a set of materials necessary for the construction of a wormarium.
3. Provide each group with the two handouts - How to Construct a Wormarium and Observation Schedule.
4. Go through the steps of construction of the wormarium demonstrating the process if necessary.
5. After the wormarium is set up, students fill it up with the soil collected.
6. The earthworms are then released into the wormarium.
7. A first observation of the behaviour of the earthworms on being released is made and then described in the Observation Schedule.
8. Students are told to set up a convenient time schedule for making observations for the next few days, at the end of which each group will be required to submit a report on their observations of the behaviour of earthworms.
9. Hold a discussion on the reports and on the importance of earthworms in the aeration of soil.

Teaching hints

The transparent glass sides of the wormarium should enable students to observe the behaviour of the earthworms inside the soil. However, it may be difficult to see them in action if there are too few earthworms inside. Therefore, ensure that there is enough worms in the wormarium so that at least some get near to the sides and can be observed.

Ensure too, that there is litter at the surface of the soil. Litter may have to be broken up into small pieces to fit into the wormarium. Do not fill the wormarium right up to the top. Leave at least 10 cm. at the top vacant. It is necessary to water the soil regularly.

Discuss with students some topics worth investigating e.g. distribution of worms, territoriality, food intake and food preference and burrow building.

Follow up

The initial observation activity should be extended to include some qualitative experimentation with the wormarium.

1. Students may be encouraged to manipulate conditions of the wormarium and observe their effects on the behaviour of the worms. Some of the conditions could be light, humidity, heat and types of soil.

2. Some investigations into the reproductive rate of earthworms are worth pursuing. Factors affecting reproductive rate can be studied.

3. Encourage students to build up an information and reference file on earthworms, such as types of earthworms, studies on earthworm behaviour, earthworms and soil fertility, and the use of earthworms as a source of food.

5. VEGETATION PROFILE OF A POND

(A, B; a, d: I, II, III)

Objectives

1. to describe the diversity of aquatic plants (macrophytes) found in a pond.
2. to construct a vegetation profile from the open water to the shoreline.
3. to relate the distribution of macrophytes to water depth, soil texture (substrate), pH, dissolved oxygen and light penetration.

Curriculum links

Aquatic environment, aquatic macrophytes, diversity indices, soil-plant relationships, sampling techniques (profile plotting).

Preparation

Teacher: Select sampling sites at suitable macrophyte-infested ponds. Provide handouts on procedures of activity. Instruct students on proper use of field instruments and laboratory techniques. Show slides or pictures of plants prior to field trip which could aid in the quick identification of plants in the field. Advise on all precautions to be taken to ensure students' safety in the field. Provide life jackets if necessary. Make certain it is safe for students to wade in the water. If not use a boat from which to use equipment or ensure students have adequate leg

covering and a life jacket or are roped to the shore. Instruct students on the methodology of computing H', the diversity index. (This could be left until they return from the field work.)

Students: Read handouts given by teacher. Revise sampling techniques. Revise use of identification keys. Revise analytical techniques and instrumentation required.

Equipment:
soilcorer
metre rule or pole marked in metric scale
raffia or string
bamboo poles
polythene bags
wet labels and pencils
pH meter
dissolved oxygen meter
secchi disc
set of soil sieves

Strategy

Question: What kind and form of plants inhabit the littoral zone of ponds?

1. Sample only the littoral zone i.e. the shallow region from the shore line to the limit of rooted vegetation.

2. Use poles (e.g. bamboo) as stakes to determine the depths at 2 metre intervals (smaller if the slope of the bed of the pond is shallow). In addition, take light penetration readings (see page 27), and pH (see page 36) and dissolved oxygen (see page 32) measurements every 2 metres. Collect substrate samples using the soil corer and place the soil in polythene bags for transfer to the laboratory. Analyse the soil texture using a set of standard soil sieves subsequently.

3. Divide the line of the stakes into quadrats of 1 metre square. This is easily done by a student holding out a metre rule on each side of him as he stands by a stake as other students in the group complete sampling the plants in the contiguous quadrats thus marked out.

4. Count the macrophytes that occur within each sample quadrat and classify as rooted but emergent, rooted and submergent, floating or submerged.

5. Measure each plant's height from the level of the substrate and its horizontal extent. Compute the average height and cover (the horizontal extent) of each species in each quadrat.

6. Construct a profile diagram of the pond littoral zone showing the water level and the substrate on a suitable scale. Mark in the sampling points, and indicate the position of the individual plant species (drawn to scale to their respective heights and spread) distributed over the transect.

Tabulate all physico-chemical data and then plot values graphically below the transect profile to indicate the environmental continuum.

Teaching Hints

1. Students are to work in groups. Groups can be assigned different responsibilities.

2. An 'environment' group can be responsible for laying the transect baseline and demarcating the 2 metres intervals; and at the same time taking the light penetration, pH and dissolved oxygen measurements and collecting substrate samples. (See Table 10)

3. Each group will need to tabulate their results on the blackboard for the rest of the class to take down. The students can then proceed to draw the profile and plot the graphs as an individual exercise.

4. A 'plant' group can be responsible for the collection of, and bringing back, the plant samples to the laboratory. For effective work, this group can be subdivided, one sub-group taking care of recording and labelling while another making height and cover measurements of all individuals within each quadrat and classifying them into respective growth froms at the same time.

Follow up

1. Discuss the interrelationships of the vegetation and the abiotic parameters of the pond along the transect. Explain the dominance of the particular macrophytes in the profile studied.

2. Explain the morphological variations/plant growth forms in relation to distance from the shore. Prepare a table showing the general adaptations of the plants inhabiting each segment of the water along the transect.

References

Butler, D. and C. A. Lawson, (1973) The Pond. Univ. California, Berkeley.

Bailey, N. T.J. (1959) Statistical Methods in Biology. English University Press.

Miles, P. M., Miles, H. B. and Graham, V. E. (1970) Tropical Freshwater Ecology. Roulton Educ. Publ., London.

TABLE 9

VEGETATION DATA SHEET

Quadrat number	Name of species	Growth & habit*	Number of individuals	Average height	Average spread	Remarks**

* state whether rooted and emergent, rooted and submergent, free-floating, submerged

** state whether vegetative, flowering or in fruit

TABLE 10

ENVIRONMENT PROFILE DATA SHEET

Distance Shoreline (metres)	Parameter
6 4 2 0	
	O_2 (ppm) surface
	O_2 bottom (ppm)
	Transparency (metres)
	Water depth (metres)
	pH
	H'

N.B. The values in the above table may be plotted below the vegetation profile.

6. ENERGY BALANCE IN ANIMALS

(B, C; I, II, III)

Objectives

1. to compute the various components of the energy balance equation:

$$E_i = E_w + E_r + E_p$$

where E_i is the energy input in the form of food intake

E_w is the energy loss in the form of excretion and faeces

E_r is the energy loss in the form of respiratory losses

E_p is the energy retained by the animal for building up tissues i.e. as secondary production.

2. to gain an understanding of energy flow through ecosystems.

Curriculum links

Community production and energy flow, trophic relationships, animal energetics.

Preparation

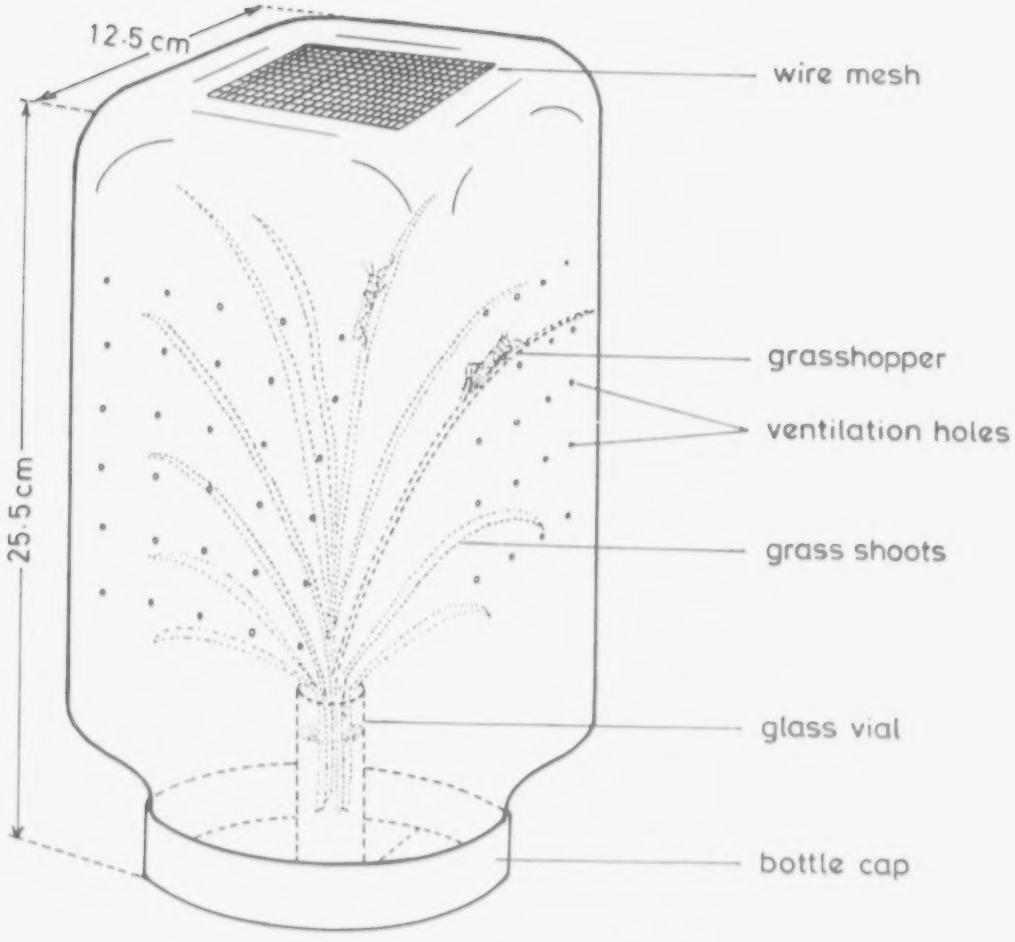
Teacher: Collect (or get students to collect) a supply of common grasshoppers from low-lying grass vegetation (see page 16). Starve the animals before the start of the experiment but do not let them die. Set up several rearing chambers from ordinary polythene sweet bottles turned bottom-up. Make several holes in the sides for ventilation and an opening with a gauze cover at the top of the inverted bottle to allow introduction of the animals or food (see Figure 44). Secure a glass vial, in the position shown, through a fitting hole made on the bottle cap. Provide handouts on procedures of activity.

Students: Read handouts given by the teacher. Read about rearing animals, especially insects, in the laboratory.

Equipment:
rearing chambers
grasshoppers (or suitable herbivorous animals if not available)
fresh supply of grass
weighing balance
weighing vials
aluminium foil

FIGURE 44

A REARING CHAMBER CONSTRUCTED FROM AN INVERTED POLYTHENE SWEET JAR



Strategy

Question: What are the energy exchanges that have to be taken into account if an energy budget or balance is to be prepared?

1. Set up the animals in groups (4 or 5) or as individuals depending upon the size of the animals in the specially prepared rearing chambers. If groups of animals are used, select those of the same age or size. Record the weight of each animal or group of animals at the beginning of the experiment.

2. Introduce weighed quantities of grass into each chamber. Since plant materials will lose water, maintain a control chamber, with only the food and without the animals, to measure the water loss.

3. Weigh the left over food in each chamber at intervals of 24 hours, introducing fresh material each time. This should coincide with the activity period of the animal. Sum all the weight losses (after correcting for moisture loss) over the experimental period to give the weight of food taken (E_i) by the animals.

4. Maintain the experiment for about a week or until the animals moults. Record the final weight of the animals. The increase in weight over the time period estimates the secondary production (E_p).

5. Weigh the faecal matter collected at the end of the period from each chamber. This estimates the component E_w.

6. To determine the E_r component estimate the respiration rate in a separate experiment. Set up the apparatus as shown. (Figure 45)

   Weigh the experimental animal(s) and then introduce into a conical flask (respirometer) containing some soda lime. The soda lime wrapped in white gauze and hung from the mouth of the flask acts as the carbon dioxide absorbent in the atmosphere of the flask. Close the flask with the rubber which should have three holes, each carrying a short length of capillary tubing. Connect one of these to one arm of the manometer, another to an inverted syringe whose plunger is positioned at the 'zero' mark and the third one to some rubber tubing with a clip to act as an outlet valve. Have another conical flask set up in a similar manner but without the animal(s). Connect it to the other arm of the manometer.

   Leave the apparatus undisturbed with the outlet valves left open for about 30 minutes to allow the respirometer to reach the temperature of the surroundings (room temperature or the temperature of the water bath).

Close the valves to commence the experiment. As oxygen is being consumed, the pressure in the flask reduces and draws the manometric fluid towards the respirometer. After about 10 minutes adjust the levels of the manometric fluid in both arms to their former positions moving the plunger down the syringe of the respirometer. Note the volume and also the time taken. Repeat twice to get an average reading. Correct the readings to SP values as follows:

$$\text{ml. Oxygen} = \underset{\text{(obs.)}}{\text{ml.O}_2} \times \frac{237\ \text{K}}{(\text{T°C} + 372)\ \text{K}} \times \frac{\text{P mm Hg (obs.)}}{760}$$

where T°C is the temperature of the water bath

p mm Hg is read from the barometer

Record the oxygen consumption.

7. From the measurements on food intake and utilisation, calculate the ecological efficiencies of energy transfer based on ratios of yield to input viz.

$$\text{Assimilation Efficiency (\%)} = \frac{\text{(wt. of food ingested - wt. of faecal matter)}}{\text{wt. of food ingested}} \times 100$$

$$= \frac{(E_i - E_w)}{E_i} \times 100$$

or

$$\text{Production (\%)} = \frac{\text{(wt. of animal at time } t_1 - \text{wt. of animal at time } t_0)}{\text{total wt. of food assimilated from } t_0 \text{ to } t_1.} \times 100$$

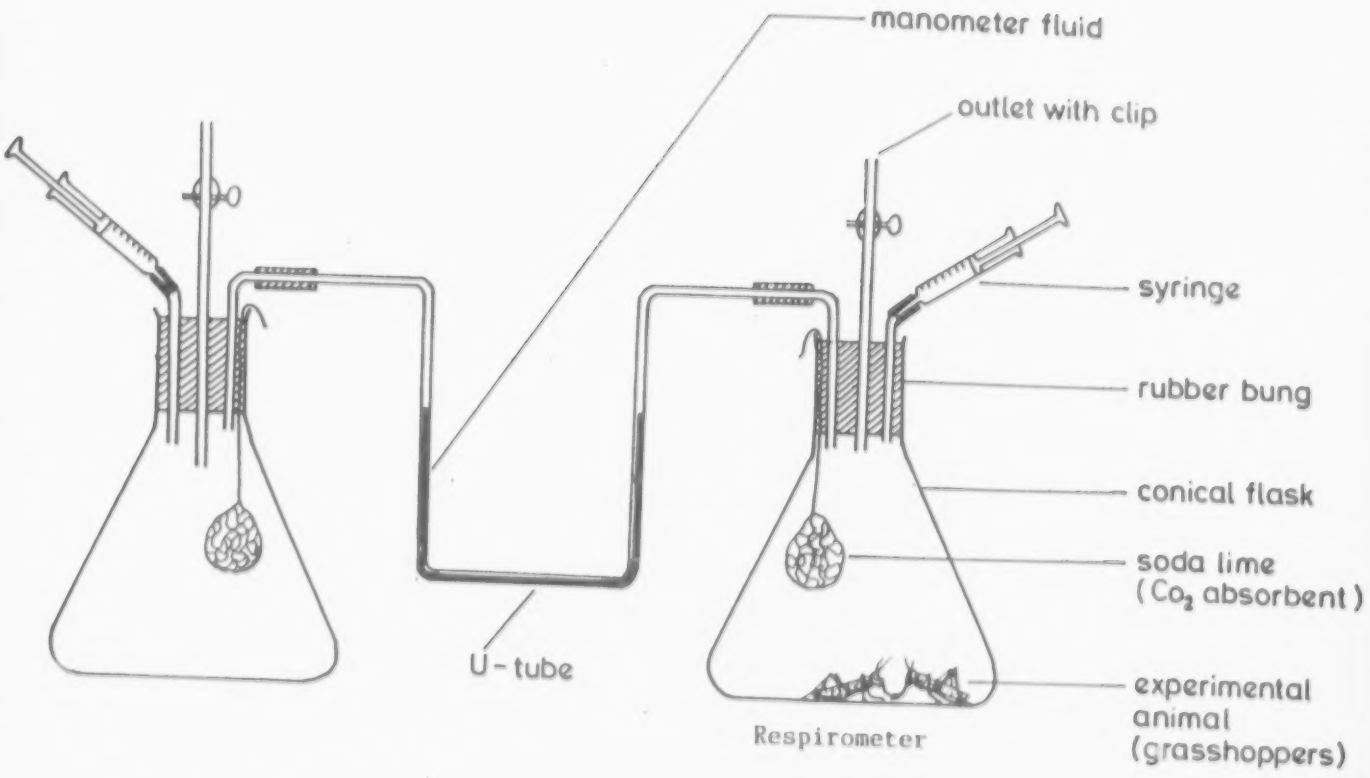
$$= \frac{E_p}{E_i - E_w} \times 100$$

8. Discuss the limitations of the experimental method.

9. Using the results of the experiment as a starting point develop the idea of energy flow through an ecosystem.

FIGURE 45

APPARATUS FOR MEASURING RESPIRATION RATE



Follow up

1. Repeat the activity using other herbivores and adapting the apparatus to them.

2. Construct simple diagrams illustratingenergy flow through ecosystems.

3. In this activity the various components in the above equation are estimated in terms of dry weight or amount of oxygen consumed. The data would be more reliable if they could be expressed in terms of energy values i.e. calories. The oxygen consumed may be converted to its caloric equivalent by mean of an **oxycaloric equivalent.** For respiratory quotient (RQ) values in the range 0.71 -1.00 (herbivores tend toe xhibit higher values than carnivores,reflecting the nature of the fuel being consumed), the energy equivalent various about 7% (i.e. 4.69 - 5.05 cal/ml of osygen).

   Determination of caloric values for food materials, animal bodies, faecal matter etc is accomplished by combustion in a bomb calorimeter. The heat released from complete oxidation is expressed as the caloric content of the material combusted, and this is taken to be its energy equivalent.

   Refer to standard books on methods of calorimetry.

References

Golley, F. B. (1961) Energy values of ecological materials. Ecology, 42, pages 581 - 584.

Kozlovsky, O. G. (1968) A critical evaluation of the trophic level concept: I. Ecological Efficiencies Ecology, 42, pages 48 - 60.

Michael, P. (1871) Energy balance in Periplanata americana L. Comp. Biochem. Physiol., 38, pages 449 - 455.

Phillipson, J. (1966) Ecological Energetics. St. Martin's Press, New York. 57 pp.

7. SECONDARY SUCCESSION ON ABANDONED LAND

(A, B, C; a, b, d: I, III, IV)

Objectives

1. to describe the process of secondary succession on abandoned land.
2. to understand the characteristics and role of different plant communities in succession.
3. to acquire practical skills in the use of line transect and quadrat sampling techniques for the study of the percentage frequency and density of plants in a particular habitat.
4. to understand the changes in plant communities as an effect in land use.

Curiculum links

Diversity and distribution, ecological balance, adaptation, colonisation, conservation, soil management.

Preparation

Teacher: Select an appropriate study site which is undergoing secondary succession e.g. abandoned crop land, mining land, grazing land or wasteland. Survey the area chosen and obtain preliminary information about its vegetation. If necessary obtain permission from the relevant authorities or landowners to study site. Obtain permission from school authorities and parents to conduct the field study. Prepare a time schedule of work. Make the necessary travel arrangements for the trip. Obtain or prepare sufficient copies of identification keys of local plants relevant to those found on the selected study site. Obtain books on local plants for use as reference. Prepare Record Sheets for students as shown in Tables 11 and 12 (pages 189 and 190). Assemble all the equipment required.

Students: Revise knowledge of local plants and soil commonly found in the selected study site.

Equipment: (for each group)
several pieces of string each of approximately 12 metres long
1 tape measure or metre stick
short pieces of wood to be used as stakes

Strategy

Question: What is the patternof secondary succession on the abandoned land? What are the percentage frequency and density of the plants found in the area?

1. Divide the students into groups (not more than 4 - 5 per group).

2. Each group is provided with the necessary equipment and Record Sheets.

3. Give instructions to all groups on procedures of setting up line transect and quadrat sampling techniques. Demonstrate, if possible, the techniques using the following outlines.

Line Transect Sampling Technique

- the line transect is used to provide an estimate of the frequency of each species of plants.

- to set up a line transect, a starting point is randomly determined such as by throwing a stone across the shoulder with the back towards the study area.

- a piece of string is run from the starting point in a random direction. The general direction of the line may be predetermined, for example, north, south, east, west (or any combination of these) from the starting point.

- the piece of string stretched between the starting and end points is secured with stakes.

- the line is marked in 1 metre lengths. There should be 10 sections of 1 metre each for the line in this activity.

- after the line is marked, the group should identify the plants touched by the line and which are up to 1 metre tall.

- observations are recorded separately for each 1 metre section. It is not necessary to count the plants; just record their presence along the 1-metre section of the line. The presence of any species along the 1-metre section, irrespective of its number, is given one tally on the Record Sheet for Line Transect Sampling. (Table II page 189) All shrubs, herbs, grasses and ground plants are to be included.

- plants which cannot be identified at the site are collected for identification in the laboratory.

- 4 more line transects are set up and observations of plants along the 40 1-metre sections of the lines are to be tallied.

- in this activity observations are made along 40 1-metre sections. The number of 1-metre sections may be reduced or increased if desired, depending, for example, on the size of the class.

- the percentage frequency of each species is calculated based on the class total tallies and the total number of 1-metre sections sampled. The formula provided in the Record Sheet (Table 11 page 189) is used.

- each group makes observations of the physical and biological features of the habitat in the area. In particular, a description of the characteristics of the dominant species and of their distributions is made and recorded.

Quadrat Sampling Technique

- the quadrat sampling technique is used to estimate the density of woody plants taller than 1 metre.

- if there are only a few, a count can be made of all the plants. Measure the area. The density is calculated by dividing the number of plants by the area.

- if there are many plants taller than 1-metre, the density is estimated by the quadrant sampling technique.

- to set up a 10-metre square quadrat, a starting corner is determined, randomly if appropriate. The other 3 corners are then set up, each being 10 metres from the other. Mark these with stakes.

- a piece of strong string is tied to the stakes to produce a square quadrat.

- identify the woody plants taller than 1-metre found inside the quadrat and count the number of individuals or each species.

- record the data in the Record Sheet for Quadrat Sampling (Table 12 page 190) provided.

- the quadrat sampling is to be repeated in different locations within the study area. The number of times it is done depends on the distribution and density of woody plants in the area.

- the percentage frequency of each species of woody plants is calculated based on the class total and the total number of quadrats sampled by the class using the formulae provided in the Record Sheet i.e. $\left(\frac{X}{Y} \times 100\right)$

- the density of the species is calculated based on the total number of plants found by the class in all the quadrats sampled and the total number of quadrats sampled by the class using the formula provided in the Record Sheet i.e. $\frac{(Z)}{Y}$.

4. Each group identifies collected specimens in the laboratory and completes all calculations in a report of the field activity.

5. Discuss findings of the field study with respect to:

   Species of plants in habitat - numbers, types and characteristics.

   Percentage Frequency of each species.

   Density of woody plants.

   Adaptability of the species to the habitat.

   Colonisation and stage of secondary succession.

   Probable climax community.

Teaching hints

1. Students should be appropriately dressed for the field activity.

2. Briefing of students should be done in class prior to the activity.

3. A herbarium collection can be prepared of each species of plant found in the study site to facilitate discussion.

4. Students should avoid trampling the study area.

5. A study should be made of the study area in relation to plant distribution before sampling begins.

Follow up

1. Sampling can be done on different localities to compare different types of succession in various habitats.

2. Investigations of soil and other physical factors e.g. light, temperature, humidity can be carried out to study their effects on plant growth and distribution.

3. Discussion on the effects of land use on changes in plant communities.

4. Locate any research studies on succession e.g. forest succession, and discuss methods and results of study with the class.

5. The same study can be replicated after a period of time e.g. six months or as a long-term project to compare different stagesof primary and secondary succession on abandoned land.

TABLE 11

RECORD SHEET FOR LINE TRANSECT SAMPLING OF HERBACEOUS PLANTS LESS THAN 1 METRE IN HEIGHT

Species	Characteristics and Distribution	Presence of Species in 1-Metre Sections		Number of 1-Metre Sections Sampled by Class (Y)	Percentage Frequency $(\frac{X}{Y} \times 100)$
		Group Data	Class Total (X)		

TABLE 12

RECORD SHEET FOR QUADRAT SAMPLING OF WOODY PLANTS 1 METRE IN HEIGHT

Species	Characteristics and Distribution	Number in Quadrat						Number of Quadrats Species is Found		Number of Quadrats Sampled by Class (Y)	Total Number of Plants in $\frac{Y}{}$ (Z)	Percentage Frequency $\frac{Z}{Y} \times 100$	Density $\frac{Z}{Y}$
		1	2	3	4	5	6	Group Data	Class Total (X)				

8. EFFECTS OF OVERCROWDING IN A PLANT POPULATION

(B, C; a, d; II, III)

Objectives

1. to understand that intraspecific competiton becomes increasingly intense as a population grows whence self-regulatory mechanisms come into operation.
2. to describe the effects of overcrowding.
3. to formulate an equation to express the relationship between the density of survivors and their total biomass.

Curriculum links

Competition, population growth, alleleopathy, regression analysis.

Preparation

Teacher: Prepare several pots of garden soil to be seeded with sunflower (Helianthus sp.) at stocking density of 50 per pot at the 16th, 12th, 8th, 4th and 2nd week before the practical. When seeding the pots, sow evenly. Provide five replicates at each age interval. Place the pots close together in the laboratory by a window, or just outside the laboratory but protected from direct sun. Randomise the position of the pots weekly to minimise edge effects. Alternately, the pots may be positioned in a 5 x 5 Latin Square formation. Provide handouts on procedures of activity. Instruct students on computation techniques viz. regression analysis.

Students: Participate in the maintenance of the pots of sunflower plants i.e. watering them regularly, removing pests and weeds. Read handouts given by teacher. Revise techniques of regression analysis.

Equipment:
large pots or black polythene bags with holes
rich garden soil
sunflower seeds
secateurs
paper bags
labels
oven
weighing balance

Strategy

Question: Which growth characteristics in plants show the greatest response to crowding (density increase)?

1. Harvest the sunflower plants of different ages.-

2. Carefully loosen the soil in each pot and remove the plants. Record the number of surviving plants (from an original 50) and the stem and root lengths of the plants in each pot. When measuring length of the stem take the height from the base at the level of the soil to the shoot apex whilst for roots take the maximum root length achieved.

3. Place whole plants in brown paper bags for further study. Ensure that these are properly labelled indicating the age of the plants.

4. Separate the plant materials into above ground and below ground portions. Place them in an oven at 60 - 70°C. After about 4 days remove and weigh the dried material to obtain the total plant biomass.

5. Record all data on your data sheets. Calculate the mean values for the following parameters for each pot: mean stem length, mean root length, mean total plant length, mean stem (above ground) dry weight, mean root (below ground) dry weight, and mean plant (total) dry weight.

6. Graph the results against the number of surviving plants separately for each growth characteristic viz.

   stem length vs no. of survivors/pot
   root length vs no. of survivors/pot
   total length vs no. of survivors/pot
   stem weight vs no. of survivors/pot
   root weight vs no. of survivors/pot
   total weight vs no. of survivors/pot

7. Take the logarithmic values of the parameters (for both x-axis and y- axis) when plotting. The gradient of the straight line in these log/log plots is given by the regression coefficient:

$$b = \left(\sum xy \quad \frac{(\sum x)\ (\sum y)}{n} \right) \Big/ \sum x^2 \quad \frac{(\sum x)^2}{n}$$

   where for instance,

   y = log mean total dry weight per surviving plant
   x = log density for surviving plants.

8. Compute b for the relationship between density of survivors and their total biomass (dry weight) by substituting the analogues into the formula. Then compute a from the formula:

$$a = \bar{y} - b\bar{x} .$$

The regression equation is given by

$$\bar{y} = a + b\bar{x}$$

[For elementary students stages 7 and 8 can be omitted.]

Teaching hints

1. When starting the cultures select very quick growing plants e.g. Helianthus sp. or Brassica sp., commercial cultivars that give rapid and even germination.

2. Instead of starting from a fixed density and then allowing self-thinning to occur in the pots, vary the number of seeds you sow per pot e.g. 2, 4, 8, 16, 32 etc.

3. The effect on density on plant biomass can be similarly obtained.

3. Always have more than the number of pots required planted out to allow for failures (viz. some seeds fail to germinate).

4. When plotting the graphs in stage 7, read the logarithmic values of the x- and y- co-ordinates from a logarithm table.

5. Computation techniques on regression analysis are available from standard statistics textbooks or micro-computer programs.

6. The effect of crowding manifests itself in a number of growth parameters. In particular, there is a constant relationship between the density of survivors and their total biomass, often, known as the power law (see White and Harper, 1970). This can be discussed further with more advanced students. Graphs of mean dry weight per surviving plant against density of survivors from self-thinning experiments (see Figure 46) in a number of species, expressed as log/log plots, exhibit a gradient of -1.5 within the 5% confidence limits. Thus if the computed b value falls between -1.25 and -1.83, it means that the results conform to this general relationship.

   Try the same experiment for different species other then the ones suggested here to test the universality of the power law.

Follow up

Discuss the roles of density dependent factors and negative feedback in the self regulation of a population. Draw on supporting materials from the experiment carried out.

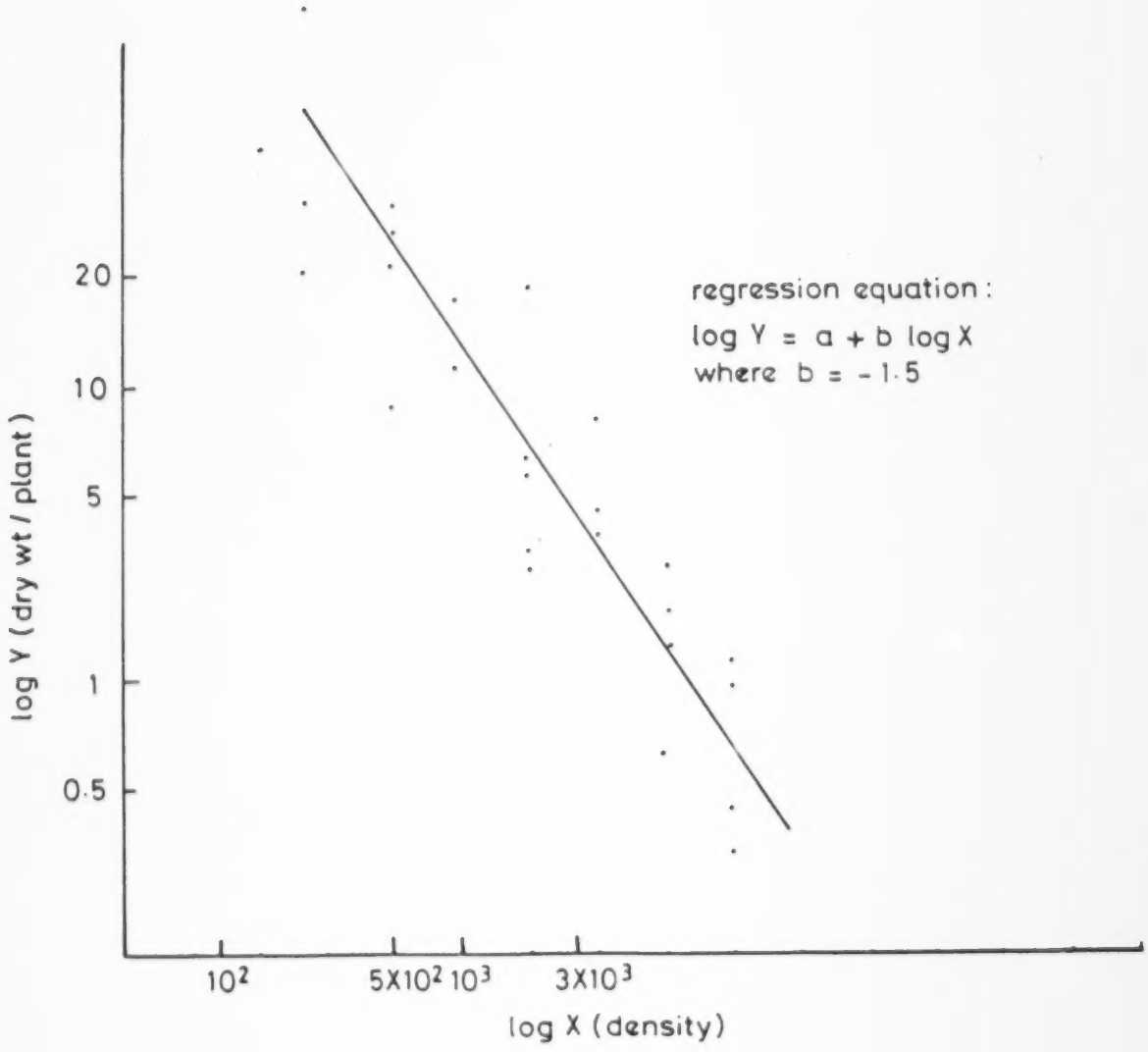
References

White, J. and Harper, J. L. (1970) Correlated changes in plant size and number in plant populations. J. Ecology, 58, pages 467 - 485.

Yoda, K., Kira, T., Ogaw, H. and Hozumi, H. (1963) Self thinning in overcrowded pure stands under cultivated and natural conditions. J. Biol. Osaka City Univ., 14, pages 107 - 129.

FIGURE 46

GRAPH OF MEAN DRY WEIGHT PER SURVIVING PLANT/DENSITY OF SURVIVORS



9. EFFECTS OF CROWDING ON AN ANIMAL POPULATION

(A, B, C; d; I, II, III)

Objectives

1. to describe the effects of crowding on the growth rates of tadpoles.
2. to isolate the inhibitory principle ('algal-like' cells sensu Richards, 1958) from 'crowded' or conditioned water and test for intra- and inter-specific effects on a number of species.

Curriculum links

Inhibition, feedback mechanisms in population growth, competition, metamorphosis.

Preparation

Teacher: Obtain frog or toad tadpoles by collecting egg masses from breeding pools or ponds. Maintain each egg mass separately and culture till the tadpoles hatch. Use filtered pond water or collect rain water forall cultures. Condition some water for the later part of the experiment viz maintain about 12 (vary the number if necessary but the total weight should be the same) large tadpoles in several one-litre portions of water contained separately in finger bowls for two days on minimal amounts of food (boiled lettuce or similar greens). Remove the tadpoles at the end of the period. Clear the water of left-over food to avoid contamination but allow the faecal matter to remain. The 'crowded' water is ready for use. Provide handouts on procedures of activity.

Students: Read handouts prepared by the teacher. Watch over the hatching of the frogs eggs. Assist in maintaining tadpole cultures to produce a supply of 'crowded' water for the experiments. Read about techniques on rearing tadpoles. Revise notes on metamorphosis of tadpoles.

Equipment:
finger bowls (18 cm. diameter)
tadpoles
rain water(or filtered pond water)
scoop nets
weighing balance
microscope
vegetables (lettuce, cabbage or spinach)

Strategy

Question: To what extent does the number of tadpoles affect their survival through to metamorphosis? What factors influence survival?

1. Set up a series of finger bowls each containing about 1 litre of fresh rain or filtered pond water under identical light and temperature conditions. Introduce into the bowls the following number of tadpoles in replicates: 1, 2, 3, 4, 5, 6, 7, 8,16, 32, 64. Take tadpoles of similar size and weight, and from a single egg mass. The latter condition ensures that inherent genetic differences which may influence the comparative growth rates are minimised.

2. Provide the tadpoles with plenty of food and change the culture water every two days. Observe the growth of the tadpoles and record, if any, deaths or metamorphoses that take place. Weigh the tadpoles periodically. To weigh, lightly blot the tadpole on a filter paper to remove excess moisture. Weigh quickly and avoid rough handling.

3. At the end of three weeks, compute the total weight of tadpoles obtained in each finger bowl. Compute the average weight per tadpole, the average weight of the tadpole before it absorbed its tail, and the weight of the largest and the smallest tadpoles. Relate to crowding i.e. the increasing number of tadpoles that were reared together.

4. In a separate experiment, set up another series of finger bowls but this time fill them up with conditioned water (containing faecal matter of tadpoles that had been reared in it for two days). Into each of these bowls introduce four fresh tadpoles of the same species that were used for crowding the water, or of a different species. For control, maintain four tadpoles in fresh i.e. unconditioned water, in another finger bowl.

5. Provide as before plenty of food and renew the culture water (conditioned and unconditioned) every two days. Conclude the experiment after 12 days.

6. At the end of the period, weigh the tadpoles, and apply significant tests for weight differences observed for the treatments.

7. Discuss the results.

Teaching hints

1. In the first experiment on crowding it will be apparent that the total growth increment increases as the number of individuals is increased, and at the same time the average mass of the individual tadpole decreases. Since excess food was available at all times in the set-up, inhibition is not likely to be exercised through preemption of food. Vary the experimental conditions to test for other possible factors.

2. Assaying the culture water for its effect on growth of tadpoles (the second experiment) suggests that the inhibition is through the addition of inhibitory materials into the water by the growing tadpoles. Ask the students to examine under a microscope the faecal material of the tadpoles used for conditioning the water and discuss their findings.

3. If fertilised eggs cannot be collected from the breeding pool sites, induce mature female frogs to ovulate with pituitary injections following the method of Rugh (1934) and then artificially fertilise the ova.

   First select a few mature female frogs, pith and decapitate them. Expose the anterior pituitary and remove with a pair of forceps. Homogenise the pituitaries in distilled water.

   Hold the recipient mature female firmly and inject the pituitary homogenate into the dorsal lymph sac by the syringe piercing between the thigh and the back. If the injection is successful, the cloaca of the female will become red and slightly swollen. Twenty four hours after administration of the hormone, subject the female to 'stripping'. Hold the frog firmly in the left hand in such a way that the frog will remain immobile. Apply gentle pressure on the abdomen with the right hand and continue squeezing in the direction of the cloaca forcing out the eggs from the uteri directly into a suspension of active sperms.

   An active sperm suspension may be prepared by macerating in 100 ml. of 10% Holtfreter's solution the testis removed from a mature male frog previously treated with pituitary homogenate. Leave the eggs in the sperm suspension for a few hours to allow fertilisation to take place. Wash and distribute into rearing bowls containing 10% Holtfret's solution. (A stock solution of Holtfreter's solution is prepared by dissolving in 1 litre of distilled water, 3.5 g. sodium chloride, 0.05 g. potassium chloride, 0.10 g. clacium chloride and 0.02 g. sodium bicarbonate.)

   Teachers should only undertake artificial fertilisation after training by an expert.

Follow up

What is the ecological significance of crowding and growth inhibition effects in tadpoles growing from egg masses?

How would this lead to more efficient utilisation of resources? Would staggered metamorphosis have the same results?

References

Licht, L. E. (1967) Growth inhibition in crowded tadpoles: intraspecific and interspecific effects. Ecology, 48, pages 736 - 475.

Richards, C. M. (1962) The control of tadpole growth by alga-like cells. Physiol. Zool., 35, pages 285 - 296.

Rose, S. M. (1960) A feedback mechanism of growth control in tadpoles. Ecology, 41, pages 188 - 199.

Rugh, R. (1934) Induced ovulation and artificial fertilisation in the frog. Biol. Bull., 66, pages 22 - 29.

10. BIRD WATCHING

(A, B, D; d; I, II)

Objectives

1. to know the different kinds of birds found in different locations.
2. to be able to identify and classify birds using the relevant keys.
3. to study the behaviour and food habitats of selected birds.

Curriculum links

Food habitats, behaviour, adaptation, conservation.

Preparation

Teacher: Obtain permission from the school and education authorities for field activity. Obtain permission from parents. Determine the different location(s) for bird watching e.g. school grounds, rice field, forest fringe, urban community, mangrove swamp, lake. Prepare a work/travel plan for the students. Make travel arrangements and if party is staying overnight, make accommodation arrangements. Prepare observation schedule. Visit (if possible) the selected location(s) and prepare map. Assemble the necessary equipment.

Students: To understand identification, use of key, use of binoculars, basic knowledge of birds, to wear suitable clothing.

Equipment: binoculars
camping equipment (if overnight stay is planned)
simple identification keys of common birds
camera and tape recorder (optional)

Strategy

Question: How are different types of birds adapted to their habitats?

1. Brief students concerning the objectives of activity, work expectations and location(s).

2. Divide students into groups (not more than 4 per group) and site groups (and individuals within a group) in strategic positions in the location(s).

3. The groups are provided with identification keys of common birds and an observation schedule is provided for each student.

4. Students are taken to the location(s) early in the morning and the activity should last about 2 hours during which the observations are to be recorded.

5. If time permits repeat the activity at other locations.

6. Groups prepare short reports about their findings and present them to whole class.

7. Discussion of findings concerning the types, behaviour and food habits of birds in different locations leading to a deeper understanding of the diversity of birds and their adaption.

Teaching hints

Students should wear clothes and head gear with colours which blend into the background of the habitat. Students should be encouraged to be patient and quiet. Movements should be minimised so as not to scare the birds away. In cases where there are large numbers of birds, a few birds can be selected for detailed observation of specific types of behaviour such as courtship, flight and feeding. (See Observation Schedule, Table 13)

Follow up

A display of student reports and of photographs taken during the trips. Further discussions on birds which are not easily encountered by the students e.g. coastal or riverine birds in schools in the interior, based on publication. Study of beaks and legs of birds and the relationship of these with their habitats (adaptation).

TABLE 13

OBSERVATION SCHEDULE FOR BIRD WATCHING

Name of bird (or main features)	Time of observation	Location of bird	Description of Activities			
			Feeding (type of food)	Flying (flapping, gliding) etc.	Relating to other birds	Any other activities

11. ACTIVITY BUDGET OF A YOUNG PRIMATE

Objectives

1. to obtain experience in ethological techniques.
2. to analyse the variety of activities of a primate over a period of time.
3. to relate the behaviour of an animal to survival in its habitat.

Curriculum links

Behaviour, ecological niche.

Preparation

Teacher: Whilst ideally this activity should be undertaken in the wild this is seldom possible. A zoo or game park is a more likely site. If primates are not available other mammals could be substituted. It is best to observe only one animal at a time. A young one will be active but avoid those that undertake large and very swift movements.

Prepare coding sheets (page 205) and lists of codes (page 206).

Students: Review concept of activity budget.

The manner in which an animal spends its time will largely determine its eventual reproductive success and survival. For example, during the day a bird may spend 42% of its time foraging (i.e. searching for and eating prey), 31% resting and preening (i.e. in body care), 11% in territorial display and defence, 6% in feeding young, 5% alert for predators and 5% in social behaviour, including courtship. But slight changes in these proportions could profoundly affect its ability to forage competitively, to defend its territory effectively, to protect itself from predators, etc.

The exact proportions of time allocated to the various activities within an animal's repertoire of behaviour will vary from day to day, season to season and year to year. They will also vary with the individual, its age and sex class and its species. Nonetheless, each individual, age-sex class and species has a typical budget that differs from those of other individuals, age-sex classes and species. The typical budgets for age-sex classes and species are the most useful

in quantitative studies because they have the greatest generality---one can speak of, for example, the budget of juvenile males. Indeed, a species' budget may be considered as defining its **ECOLOGICAL NICHE** more than any other aspect of its biology.

An activity budget, therefore, refers to proportions of the day, season or year that an individual, class or species spends in all of its activities. Since it is highly impractical, if not impossible, to observe an animal all day, budgets are based on **REGULAR** and **BALANCED** samples. This may not be possible to achieve in this activity but one should attempt to get as close to it as possible.

Review data collecting method

The data-collecting method we will be using consists simply of recording numbers for the activities and other data onto **CODING SHEETS**. A coding sheet is a form for receiving numbers that can be analysed efficiently by hand or, by computer. It can be in the form of a sheet of paper or computer (IBM) cards.

The chief problem with the technique is that every bit of data (with the exceptions of headings and other non-quantifiable information) must be represented by a number. Therefore, you must have a list of **CODES** (numbers) for each type of data---that is, a list of numbers for all the activities, another list for all the identities of the animals, another for their locations, etc. The lists do not have to be complete before observations begin; new items may be added to the lists in the field.

Equipment:

writing surface (e.g. clipboard)
pencil and eraser---a pen will not do
coding sheets
list of codes
binoculars (if available)
watch

Strategy

1. Work in groups of three: all three observers will watch the behaviour of the subject, but one observer will record the data, one will consult the codes and one will note the times.

2. Select a subject.

3. Spend about an hour or so observing the subject. Identify its activities. using codes provided or creating new ones, and provide a brief description of each.

4. Review these activities and other listed in provided codes.

5. Fill out the top of the coding sheet:

   A. Date
   B. Page number (note that four pages will be needed to record observations every minute for two hours).
   C. Subject's name.
   D. Group size of the subjects.
   E. Observers' names.
   F. Weather (be brief: e.g. whether wet or not, cloudy or sunny, etc.).

6. Fill in the starting time (e.g. 11.01). [Although there are places for many times, you only need to put in the starting time and later the stopping time.]

7. Identify the following behaviour that occurs at **PRECISELY** one minute intervals (i.e. what is happening exactly at 0 seconds each minute):

   A. Subject's behaviour.
   B. Identity of any animal interacting with the subject.
   C. Type of object being handled (if any).

8. Watch for one hour. If you miss an observation, **DO NOT GUESS** what the animal was doing. But put a '?' in the activitiy box. Do not stop and re-start observations within the hour. If one minute intervals are too frequent, use two minute intervals (but not three or four, if possible)---if you change intervals, you must start from the beginning.

9. If the behaviour, object or interactant is not on the list, add it to the next unused number.

10. Fill in the stop time when the second hour is up.

11. Repeat with another subject.

Analysis

This practical will have little validity as a basis for quantitatively comparing the subjects. Not only are the samples not balanced (primarily to control for variations in behaviour correlatedwith time of day), but they are too few for meaningful comparisons.

After collecting the data, add up the frequencies of each activity, each object and each interactant. You should find that some activities are common in both subjects, some are common in one but rare in the other and many are rare in both. In the first two cases, activities are easily compared statistically and intuitively. In the last case, neither statistical nor

intuitive comparisons are valid because differences or similarities may be the results of sampling errors. Consequently, in studies with many activities, there is usually an initial lumping of activities into five or six categories e.g. placing activities such as grooming, play, bathing, etc. into the category of 'maintenance behaviour'.

Add up the frequencies for each category of each subject. With more advanced students analyse for statistically different budgets using chi-square analysis.

The next step is to examine each category in turn. Each could be compared statistically between subjects by binomial tests. Draw conclusions (without statistics) about the similarities and differences between the subjects. Refer back to frequencies of individual activities that compose the categories.

Briefly examine the frequencies of object use and of interactants.

Discuss the relation between the animal's behaviour and its habitat.

Follow up

Discuss issues concerned with the validity of sampling methods and analyses.

A CODING SHEET

Date ..... Page .... of .... Pages

Subject's name .....

Observer's names .....

Weather .....

OBSERVATIONS	Time				Activity	Object	Inter-actant								Remarks
1															
2															
3															
4															
5															
6															
7															
8															
9															
10															
11															
12															
13															
14															
15															
16															
17															
18															
19															
20															
21															
22															
23															
24															
25															
26															
27															
28															
29															
30															
⊠	1	2	3	4	5	6	7	8	9	10	11	12	13	14	15	

EXAMPLES OF CODES FOR ACTIVITIES

? = unknown (subject is out of sight or activity can't be identified
2 = ventral riding.
3 = quadrupedal locomotion (walking with hands and legs).
4 = bipedal locomotion.
5 = brachiation (moving by swinging the arms from one hold to another).
6 = hanging (by arms and without moving).
7 = standing (if grasping object identify in the appropriate column).
8 = climbing (on rocks or poles, or climbing in general).
9 = cradled by mother (or father).
10 = clinging (to interactant---identify which animal).
11 = manipulating object (identify).
12 = wrestling or playing (identify interactant, if any).
13 = breast-feeding.
14 = eating solid food (self-feeding).
15 = withdrawal from another animal (identify).
16 = somersaulting.
17 = inactive while sitting alone.
18 = drinking.
19 = swinging.
20 = biting.
21 = digit sucking.
22 = sleeping or resting.
23 = lying.
24 = playing.
25 = squatting.
26 = urinating.
27 = exploration.
28 = scratching ground.
29 = holding objects while supported.
30 = sliding.
31 = bending.
32 = spider-walking.
33 = holding object while not supported.
34 = huddling (tight grouping with other animals).
35 = shattering.
36 = yawning.
37 = grooming baby.
38 =
39 =
40 =

EXAMPLES OF CODES FOR OBJECTS AND INTERACTANTS

OBJECTS		INTERACTANT	
?	= unkown.	?	= unknown.
1	= food.	1	= Mother.
2	= stick.	2	= Father.
3	= leaf or leaves.	3	= Uncle.
4	= vine.	4	= Aunt.
5	= paper.	5	= Young unrelated male.
6	= rock.	6	= old unrelated male.
7	= chain.	7	= young unrelated female.
8	= rail.	8	= old unrelated female.
9	= stump.	9	= baby
10	= plastic.	10	=
11	= central pillar.	11	=
12	= tyre.	12	=
13	= coconut husk.	13	=
14	= bar.	14	=
15	= fruit skin.	15	=
16	= tin		
17	= foil.		
18	=		
19	=		
20	=		

12. LOCOMOTION AND RESPONSES OF PLANARIA

(A, D; I, II, III)

Objectives

1. to observe the locomotory and response behaviour of a simple aquatic organism.
2. to relate the organism's behaviour to its ecological situation.
3. to undertake experimental investigations on behaviour.

Curriculum links

Behaviour, stimulus and response, taxes and kineses, ecological niche, primitive metazoa.

Preparation

Teacher: This activity is undertaken in the laboratory but should be linked to observations in the field if at all possible.

Students: Review the nature of planarians. They belong to the phylum Platyhelminthes and Class Turbellaria (free-living flatworms). Turbellarians are mostly small (never more than a few centimetres), and found in many freshwater habitats, and also in marine and humid land habitats. They are mostly carnivores or carrion-feeders. They have great powers of regeneration. Turbellarians are the most primitive metazoans (multi-celled animals) that regularly move about in search for food, shelter, mates, etc.

This activity involves four investigations concerned with locomotion, response to light, food location and taxes. It is suggested that they be undertaken in parallel by groups in a class. The groups report back to the whole class when they finish.

Locomotion: Planarians rest in the open in a contracted, rounded condition but are usually extended (and quiescent) in the angle of a Petri dish. They have two basic types of locomotion: gliding and alternately extending and contracting. **Gliding** occurs when the flatworms are submerged. They extend fully and glide forward with their heads just clear of the substrate. Their smooth movement is accomplished by cilia on the worm's undersurface. The cilia cause small currents of water as the worm moves. [The Latin word **turbella** means a small disturbance.] Gliding takes the worm smoothly over a variety of substrates under water. It is aided by the secretion of a fine mucus on which the worm travels.

Extending and contracting is most often seen when the worm is in very shallow water or out of water. Often during this type of locomotion, the worm waves its headas if searching for the best direction to move.

Equipment:

Petri dish with 6 planarians
tray of pond water, about 30 cm. x 30 cm.
sheet of clean glass, 25 cm. x 25 cm.
container of powdered talc
two cavity slides
four cover slips
low-powered stereomicroscope (x 20 or x 40)
tube of carmine powder
wide-mouth pipette
access to monocular microscope
fine soft paint brush
white tile

Strategy

Question: What determines the movement of Planaria?

1. Observe undisturbed flatworms in the Petri dish. Describe their postures at various locations in the dish.

2. Gently shake the dish. Observe under a low-power steroscope the gliding movements of the worm. Describe gliding.

3. Gently pipette a flatworm onto a cavity slide. Add a small amount of carmine and observe the action of the cilia under a monocular microscope.

4. Place a flatworm in the tray with the piece of glass on the bottom. After the worm has moved around on the glass, at least several inches, remove the glass, sprinkle it with talc and gently wash the excess talc away. There should remain a mucus track with talc adhering to it.

5. Pipette a worm onto the white tile. Vary the amount of water and describe the worm's movements.

Responses to Light

Taxis---directed movement, either away, towards or at an angle to the stimulus.

Kinesis---undirected movement in which the stimulus produces a change in speed or change in the frequency or turning.

Thus there are two mans by which an organism avoids unfavourable environments. Firstly, it may direct its movements with respect to the stimuli, or, secondly, it may change the speed of its movement or frequency of its turning depending on the strength of the stimulus. Your problem will be to determine which the planarian uses.

Equipment: Petri dish containing 10 flatworms
white tile
wide-mouth pipette
torch with narrow beam
several headless worms (that have been regenerating for 1 or 2 days)

Strategy

1. Place the lamp near the Petri dish, and observe the movements of the worms. Vary the distance of the lamp. Do the worms move directly away from the light? Or do they increase their speed or frequency of turning near the light? In other words, are they displaying taxis or kinesis?

2. Streak a dozen or so drops of water across the tile. Place the worms carefully in this streak. Then shine light at a right angle to the streak. What do the worms do? (Is their movement direct or indirect? What types of locomotion are used?.) After several minutes, turn the tile around 90 degrees and observe their behaviour. What effect does the restricted water supply have on the worms response to light?

3. Do each test again with headless worms. Do they respond? Are eyes important for phototaxis or photokinesis?

Food location

Equipment: container of 10 flatworms that have been starved for 7 days.
tray of pond water
three Petri dishes
wide mouth pipette
container with various baits (e.g. liver)
three small specimen tubes
headless and bodyless worms that have been regenerating for 1 or 2 days.

Strategy

1. Place a very small piece of liver in the tray and observe the reactions of the worms. Gently herd the worms into one corner and place meat in the other. How long does it take the first worm to react? What kind of movements do they make when first alerted? When moving toward the food?

2. Vary the amount of food given each time and observe differences in responses.

3. Place some food carefully in a specimen tube after washing your hands (to make sure that the outside of the tube has no trace of food on it). Place the tube in water near a worm, making sure the top stays out of the water, and observe its response.

4. Test food locating abilities of headless and bodyless worms. Do they differ?

Thigmotaxis and Rheotaxis

Planarians are negatively sensitive to touch (thigmotactic) on their dorsal surface, and positively thigmotactic on their ventral surface. This is true regardless of gravity---a planarian will not come to rest if both of its surfaces are in contact with substrates. Planarians also react to water currents produced by animals that they prey upon.

Equipment:
fine pipette
tubing (to fit water tap and fine pipette)
Petri dish with 10 flatworms
paint brush (fine)
deep dish (3 or 4 inches deep)

Strategy

1. Determine how planarians react to a current of water and which parts of their body are sensitive by directing a fine gentle stream from (i) directly in front of the worm, (ii) directly behind; (iii) side of body, and (iv) side of tail. Which parts are sensitive? How does the worm react?

2. Vary the strength of the current. Are responses quicker to stronger currents?

3. Test headless planarians for rheotaxis. Do they respond?

4. Drop several planarians in a dish with 3 or 4 inches of water. Which end contacts the bottom first? Do they always land right-side up?

5. Lightly touch a still worm with a paintbrush. How does it respond? Turn a whole worm over and time how long it takes to right itself. Then do the same with a headless worm. What is the likely role of the brain in the righting reaction?

6. Discuss the findings of the investigations in relation to the survival of planaria in natural habitats.

Follow up

Discuss the validity of the methods used in this investigation.

13. MAMMAL TRAPPING IN THE FOREST OR AN AGRICULTURAL PLANTATION

(A, B, C, D; a, d; I, II, III)

Objectives

1. to study the characteristics of the small mammals found in the habitat.
2. to study the relationship between the characteristics of the small mammalian population and those of the habitat.
3. to use trapping techniques in catching small mammals.
4. to handle various kinds of small mammals including marking technique for further studies.
5. to understand pest control of mammalian pests in the case of agricultural plantations.

Curiculum links

Habitat study, pest biology, conservation, techniques of trapping, handling and marking, mammal identification.

Preparation

Teacher: Select a suitable secondary forest or an agricultural plantation on the outskirt of town, preferably near a residential area. Be aware of any possible danger such as the use of poisonous vipers as predators to control rodents in oil palm plantations. Obtain permission from relevant authorities and parents for field trip. Make travel arrangements and prepare work schedule. As the site has to be visited several times it should be as close to the school as possible. Prepare the necessary equipment and identification keys.

Students: Dress appropriately particularly wth stout shoes and to have gloves available. Review knowledge of local small mammals and forest habitat.

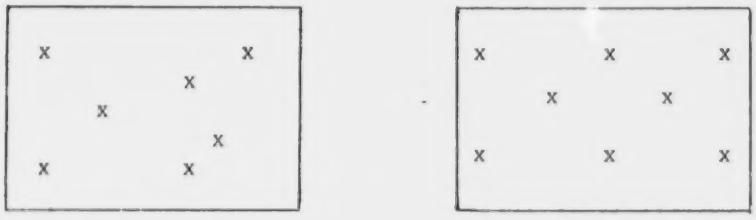
Equipment:
rodent traps
baits - local fruits
toe clipper
spring balance
poles as markers
watch
first aid kit

Strategy

Question: What are the types of small mammals found in a secondary forest or agricultural plantation?

1. Select a particular site for trapping. Depending on the size of the study area, groups may be assigned to work in specific trapping sites.

2. Within each trapping site, the randomised or uniform design could be used in setting up traps.



randomise uniform

3. Place the traps carefully in the preferred positions. Ensure that the baits are put inside the traps and the doors of the traps are open.

4. Put a numbered pole near to each trap as indicator.

5. Draw a plan of location where traps are set.

6. Examine the traps at regular time intervals. Decision regarding the time for observation will depend on the distance to study site and the availablility of time and work schedule. Observations may be made once, or more times a day. Fix the times for observation.

7. If animals are present in the traps, identify them. They must be treated carefully.

8. Wherever possible identifications should be made at the site. If not it could be done in the laboratory.

9. Record other information such as colour, length, weight, sex, juvenile/adult and other body characteristics e.g. dentition, relevant to their adaptation to the physical and biological environments.

10. Mark the animals. (See page 20) [Optional]

11. Release animals at the positions where they were caught.

12. The trapping and observations should be carried out over a number of days.

    [Recapture data may show particular patterns of movements of the animals. If time permits use the capture - recapture technique to estimate population size (see page 23).

13. Construct drawings etc. to describe the main features of the animals' habitat.

14. Discuss the range of animals found at the study sites and their relationships with the characteristics of the habitats.

Teaching hints

This activity may not yield results unless carried out over a period of time and until animals in the wild grow accustomed to the presence of the traps. Traps could be examined in the morning to identify animals trapped in the night and in the early evening to identify animals trapped in the day. The teacher's knowledge of the study site will help in the design of this activity for maximum information to be obtained. Times for observations should be consistent. Care should be exercised when handling animals collected in the wild.

Follow up

1. This activity can be carried out at different times or seasons of the year at the same site to study the changes or variations in the mammalian population.

2. Climatic data, characteristics of foliage, and other information can be gathered and related to the behaviour of the animals.

3. A more detailed literature study can be made about the behaviour and feeding habitats of the animals trapped in the study site. Appropriate books on the subjects should facilitate this work.

4. The activity can be carried out in different localities and the findings compared.

14. RESOURCE PARTITIONING AMONG FOREST FLOOR FROGS (ANURANS)

(A, B, D; a, d; I, II, III)

Objectives

1. to describe the diversity of anuran species co-existing on the forest floor habitat.
2. to compare the age composition of the various species populations.
3. to compare the diets of the various species from stomach content analysis.

Curriculum links

animal behaviour, diets, regulation of species diversity, population/community structure.

Preparation

Teacher: Conduct a survey of select forest sites e.g. near stream banks, ravine slope or ridge crest. Obtain samples of anuran representative of the selected study areas. Obtain samples of arthropods of the forest floor from pitfall traps. Identify the animals and display for viewing by students in vivaria. Provide handouts on procedures of activity. Instruct students on sampling/catching the anurans. Instruct students on dissection/stomach analysis techniques.

Students: Read handouts given by the teacher. Revise arthropod taxonomy and use of identification keys. Examine the various anuran and arthropod specimens on display. Revise dissection procedures.

Equipment:
lamps
cloth bags
specimen bottles
10% formaln/70% methylated spirit
dissection sets
Petri dishes
finger bowls
microscope

Strategy

Question: Do different species which overlap in a habitat eat different foods?

1. Conduct nocturnal (just after sunset) sorties along narrow forest trails with the help of a lamp. Catch with your bare hands any frogs/toads sighted on the trails. Sampling time should be about two to three hours moving at regulated pace along the trail.

   Only capture up to one or two frogs per student, depending on the size of the class. Do not kill rare species. Identify the specimens in the field.

   [If possible do this with some, if not all students. Otherwise teachers should make the collection.]

2. Kill and preserve the captures as soon as possible (within an hour of their capture) in 10% formalin. Inject the formalin solution with a syringe into the stomach.

3. In the laboratory group the individual animals according to their species. Take measurements of the standard snout-vent length and the body weight of all individuals before starting the dissection.

4. Dissect open the body cavity of each specimen to examine the internal organs. Classify in ages according to the

condition of the gonads viz.

ovulating females	(Mullerian tubules distended with eggs)
mature females	(with distinct ovaries)
mature males	(with distinct testes)
juveniles	(gonads indistinct and undifferentiated)

5. Next remove the stomach from the abdominal cavity. In the analysis of stomach contents, cut along the greater curvature of the stomach and empty its contents into a Petri dish by washing with a little water.

6. Identify the stomach contents with the help of identification keys or specimens on display. Identify particularly partially digested items e.g. leg fragments, broken wings, head capsule etc. Match these with intact specimens.

7. For each anuran species, collate the data (length, weight, and age) and construct appropriate histograms to indicate the composition of each species population. Compare species.

8. Tabulate the number of different food items for each stomach content sample for each species. Divide the mean value for each item by the mean total number of food items per stomach. The result is the relative proportion of a particular item (prey taxon) in the diet of a species.

9. Determine the differences in food of different species. Discuss how this is related to their behaviour and environment. To what extent is there competition between species for food.

10. (Optional for more advanced students.) To ascertain overlap in the diets (resource utilisation) of any two competing species in the same habitat, calculate the following measure of competition coefficient viz.

$$\alpha_{ij} = \frac{\sum_{1}^{m} p_{ia} \; p_{ja}}{\left(\sum_{i}^{m} p^2_{ia}\right) \left(\sum_{i}^{m} p^2_{ja}\right)}$$

where p_{ia} and p_{ja} are respectively the proportion of resource **a** in the diets of species **i** and **j**.

An example of a calculation is given in Table 13, page 218.

α_{ij} quantitifies the degree of similarity in the diets between species. Values range from 0 when species share no common prey items, to 1 when the diets of two species are identical.

Teaching hints

1. Catching anurans in the dark needs practice. Always look ahead as you move along the trail. Clip lamp (or use a helmet lamp) to clothing so as to leave hands free. Reverse reflection of the beam of light from the lamp by the animal's eyes betrays its position to you.

2. When dissecting the animal, pin out the limbs so that the belly is uppermost. Cut through the body wall to one side of the mid-ventral line from the cloaca to the jaw symphysis. Locate the liver and turn it aside to see the gut. The wide sac-like stomach lies to the left of the liver. If the contents cannot be examined immediately, split open the stomach to allow penetration of preservative and then store them in 70% methylated spirit until subsequent examination of their contents.

   To ascertain the reproductive state of the animal, displace the viscera to one side and locate the kidneys in the roof of the body cavity. Note the orange fat-bodies anterior to the gonads. In the female, the ovaries (which also lie anterior to the kidneys) are paired bodies suspended from the kidney by a mesentery (the mesovarium); the oviducts (Mullerian tublues) being a pair of long, coiled tubes in the dorsal, peritoneal lining of the body cavity. In the male, the testes are attached to the ventral surface of the kidneys by a mesentery (mesorchium); the minute vasa efferentia run from the testes in the mesorchium carrying sperm through the kidney tissue to the common urino-genital (Wolffian) duct.

3. To obtain information on the type of food items (prey) available to the anuran species, collect the forest floor dwelling arthropods using pitfall traps overa twenty-four hour period. These are glass jars, bottles or cans sunk into the soil in such a way that the mouth of the container is flush with the soil surface. (See page 53)
   Place a protective cover (e.g. a large leaf), raised from the soil surface, over the mouth. In order to prevent larger animals (predators) from killing the smaller individuals trapped together with them, introduce into the pitfall traps a preservative, like saturated aqueous solution of picric acid, at the time of setting the traps.

   Set up a number of pitfall traps in suitable locations in your sampling area. If you are carrying out a twenty-four hour survey, you can examine, empty and reset the traps at regular intervals. Sort out the species and identify them.

Follow up

What is the basis for interspecific competition in the anuran community in your study area? Discuss in relation to interference e.g. agression, territoriality and differential resource exploitation. Substantiate your answer with observed evidence.

References

Bullock, J. A. (1966) The food of amphibians and reptiles. Bull. Nat. Museum (S'pore), 34, pages 85 - 96.

Schoener, T. W. (1974) Some methods for calculating competition coefficients from resource-utilisation spectra. Am. Nat., 108, pages 332 - 34o.

Toft, C. A. (1982) Community structure of litter anurans in a tropical forest, Makokou, Gabon. Rev. Ecol. (Terre Vie)., 36, pages 223 - 232.

TABLE 13

EXAMPLE OF A CALCULATION OF COMPETITION COEFFICIENT

Resource (prey taxon)	% in the diet (p) Rana blythii (i)	Rana hosii (j)	$P_{ia}P_{ja}$	P^2_{ia}	P^2_{ja}
Orthoptera (Tettrididae) Tettigonidae adults and nymphs)	0.24	0.08	0.019	0.058	0.006
Coleoptera (Scarabaeidae, Curculionidae, Elateridae)	0.31	0.06	0.019	0.096	0.004
Hymenoptera (Mainly Formicidae)	0.23	0.61	0.140	0.053	0.372
Dictyoptera (Blattidae adults)	0	0.03	0	0	0.001
Miscellaneous arthropods	0.11	0.10	0.011	0.012	0.010
Chilopoda	0	0.05	0	0	0.003
Aranea	0.07	0.09	0.006	0.005	0.008
Diplopoda	0.04	0.07	0.003	0.002	0.005
			0.198	0.226	0.409

$$\alpha_{ij} = \frac{\sum_{i}^{m} P_{ia} P_{ja}}{\left(\sum_{1}^{m} P^2_{ia}\right)\left(\sum_{1}^{m} P^2_{ja}\right)}$$

$$= \frac{0.198}{0.226 \times 0.409} = 0.651$$

15. COMPARATIVE STUDY OF POLLUTED AND UNPOLLUTED AREAS OF A STREAM

(A, B, C; a, b, c, d; I, II, III)

Objectives

1. to describe how the physico-chemical characteristics of a stream change with pollution.
2. to compare the biological diversity of clean and polluted environments.

Curriculum links

Diversity, distribution, habitat, ecosystem, biotic community, pollution, conservation, environmental management.

Preparation

Teacher: Survey and select site. Obtain permission from owner of site for the study. Obtain permission from the school authority and parents to take out students. Prepare materials needed. Brief students on the activity.

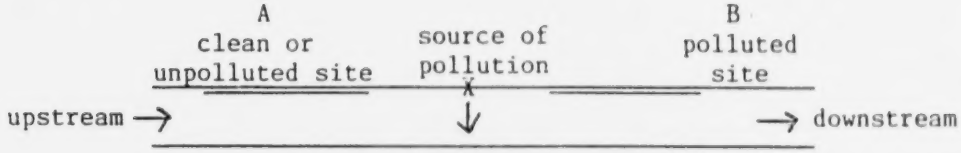
Students: Read instructional procedures before the activity. Revise concepts of ecosystem and ecological balance. Practice collecting techniques.

Equipment:
rope with weight attached
metre rule
thermometer
watch
cork or wood float
sieve, hand screen or nets (see pages
scoop
dishes
forceps
glass droppers
identification keys
materials for chemical determinations (See Chapter 4)

Strategy

1. Conduct introductory discussion on what is pollution? Identification of polluted places in the locality. Pose the question How might a polluted area compare with a clean, unpolluted site?

2. Proceed to the selected study site which should ideally be arranged as follows.



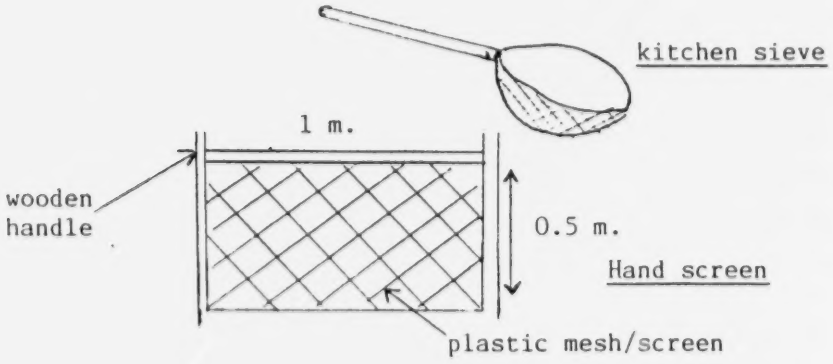
3. The following should be undertaken at both the unpolluted and polluted sites.

   (a) Students to write a general description of each site giving location and distance relative to the pollution source; vegetation in the area both on the land, and in the water; nature of substrate e.g. mud, sand, rock.

   (b) Measure some physical features of each site: stream width and depth (using a weighted rope); surface water temperature; water velocity (by noting the time taken for a float to pass along a known distance). Take three measurements in each case, average and record.

   (c) Determine some chemical characteristics of each site pH; dissolved oxygen; suspended sediments; total hardness. (Others may be appropriate according to the nature of the pollution.)

   (d) Collect macroscopic animals e.g. fish, aquatic insects, insect larvae, snails. (See previous notes on precautions needed to be taken.)



Two students work to use the hand screen. One student holds the screen in the current and the other goes upstream a few feet from the screen, turns over rocks and wipes animals off them. The current will carry the dislodged animals to the screen. In removing the screen from the water, the two handles are brought up together so that the screen forms a loop, thus keeping the animals trapped. To remove the animals that remain stuck to the screen, the screen is placed over the examination try and water is poured through the screen.
(Nets can also be used for collecting see page 16.)

Obtain samples of organisms from the bottom of the stream. A can can be used as a scoop, filled with mud and debris which is then poured into a kitchen sieve. The mud is washed away by agitating the sieve while holding it partly under water. The animals are left in the sieve.

4. Sort the collection of macroscopic animals. Transfer all the collected animals into a white enamel or plastic pan. Use a pair of forceps to pick up each one. A glass (medicine) dropper can be used to suck up very small ones. Identify and count each species. A guide to identification must be provided for each group.

5. Discuss the differences between the polluted and unpolluted sites in terms of:

   physico-chemical features of the sites

   the types and number of plant and animal species and the diversity of species

   devise methods for monitoring pollution and its effect introducing the concept of biological indicators.

SAVANNA

INTRODUCTION

The savanna biome is one of the largest of the earth's biotic systems, occupying huge areas in Australia, Africa and South America.

A savanna is a biologically complex ecosystem, with trees forming a scattered, open canopy interspersed with grasslands. Savannas occur in regions where it is tropically warm throughout the year. Seasonal rainfall is a principal controlling factor on the lives of animals and plants. Annual precipitation in the savanna averages between 50 and 150 cm and is decidely seasonal, a season of heavy rainfall alternating with drought. Towards the lower end of the rainfall range, the savanna becomes a thorn scrub ecosystem grading into desert shrublands. Towards the upper end of the range, the savanna biome grades into deciduous forests and tropical rain forest.

Vegetation South American savanna occurs on land relatively impervious to water, so that the soil alternates between being water-logged during the wet season and extremely dry during the dry season. Common trees in these savannas are Palms (*Mauritia* sp. and *Capernica* sp.). Australian savannas, which have largely disappeared under human exploitation, were characterised by open stands of eucalyptus trees (*Eucalyptus* sp.) with grass and shrub understory. The principal trees in North America savannah, which begins in Texas and sweeps southwards through Mexico, are mesquite (*Prosopis* sp.) and several acacia species (*Acacia* sp.). The African savanna covers much of that continent. Trees characteristic of the African savanna are acacias and species of *Brachystegia*, *Combretum* and *Julbernardia*. The grass cover ranges from bluestems (*Andropogon* sp.) and elephant grasses (*Pennisetum* sp.) in the wetter regions to wire grasses (*Aristida* sp.) in the drier regions along the desert margins of the savannah forming a cover only a few centimetres high. Elephant grasses occur as flanks of tropical forests and may form a dense growth 3 to 4 metres high. Red oats grass (*Themeda triandra*) is one of the most common grasses over large areas of Africa. Tree abundance varies from scattered individuals to fairly dense stands; and escarpments may be covered almost completely by woody plants.

Primary Productivity The net primary productivity of the savanna varies considerably along the gradient from thorn shrub and wire grass to acacia and elephant grass ecosystems. Productivity estimates for the above ground herbaceous layer range from 37 to 800 g/m^2 year, or on a growing season basis, 0.67 to 8.12 g/m^2 day. There are few data available on the contribution of trees to primary productivity, although they are the major food source for such browsers as giraffes and elephants. Many trees are deciduous. Some, notably *Acacia albida*, which is abundant in the northern savanna region of Africa, produce leaves during the dry season and lose them at the onset of the rains. Leaf fall enhances herb productivity underneath the trees, probably as a result of nutrient enrichment. Trees may serve as nutrient pumps in the savanna, taking up nutrients from deep within the soil by virtue of their deep root systems and enriching surface layers by leaf fall.

Consumers The savanna contains a variety of consumers. Arthropods usually have peak populations at the beginning of the dry season. Birds that feed on plant reproductive structures are most abundant during the middle of the rainy season, as are amphibians. Insectivorous mammals peak at the same time as their food source. All these trends are related to seasonal rainfall; temperature is relatively constant in the savanna biome.

Herbivores The African savanna supports a spectacular diversity of large herbivores and their predators. The variety of antelopes alone is extremely large: blesboks, dik-diks, duikers, elands, gazelles, gerenuks, klipsringers, kudus, oribis, sable antelopes, springbucks, waterbucks and wildbeests, to say nothing of non-antelope species such as zebra. This is in contrast with North American grasslands supporting only three large herbivores, or South American savanna and grasslands, mostly devoid of large herbivores.

How can such a vast array of herbivores co-exist? Six important ecological factors have been identified as separating common herbivores species.

1. Differences in diets: There is a clear separation between browsers such as giraffes and grazers such as wildbeests.

2. Animal occupancy of different vegetation areas differ: e.g. lesser kudus occur mainly in areas with dense tree and shrubs cover, while Grant's gazelles are found commonly in open grassland.

   Species generally similar in feeding habits and overall vegetation occupancy are further separated by -

3. occupying different areas during the same season; and

4. occupying the same area at different seasons.

   Species in all of the above potential patterns of habitat separation, such as impalas and giraffes, tend to -

5. utilise different foliage levels and

6. different plant parts.

Predators The African savanna is famous for its major predators: lions, cheetahs, cape hunting dogs and hyenas. These carnivores follow huge game herds across the savanna and are differentiated by their prey. Lions kill all of the antelopes up to such large species as the eland. At the opposite end of the spectrum, jackals either scavenge the remains of carcasses killed by one of the large predators or prey on small, immature immature ungulates.

The Web of Life in the Savanna

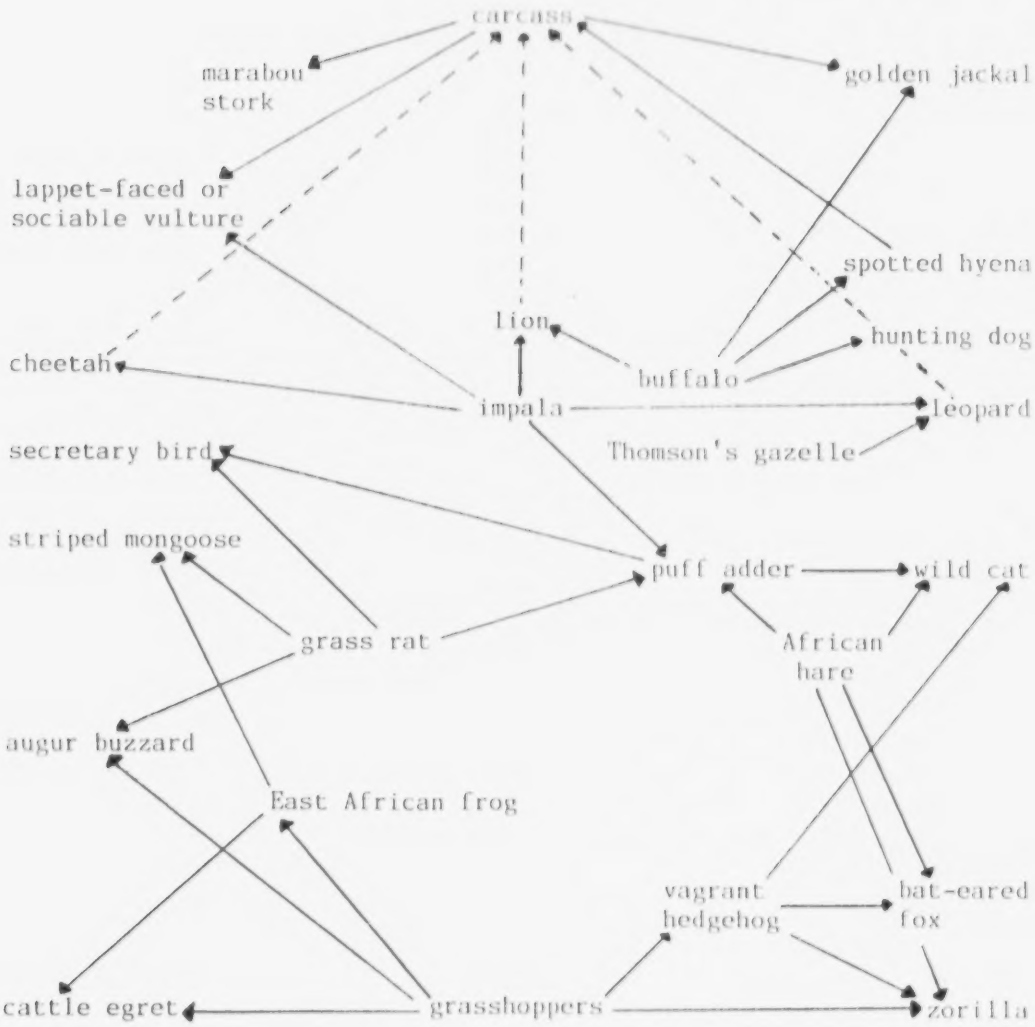
Figure 47 provides an East African example of the complex nature of the relationship between carnivores and herbivores in a savanna. Grasshoppers, grass-eating rodents and plain game, large and small, are

the three bases of the three great food pyramids of the savanna plains. In complex natural communities very few food chains are simple, very few animals avoid falling prey to other animals. Most herbivores are preyed on by either insectivores or by herbivores. Carnivores may eat insectivores, and carnivores may in turn be eaten by other carnivores. Only the great super-predators and scavengers at the summit of the pyramid are free from the threat of predation.

FIGURE 47

THE WEB OF LIFE IN SAVANNA IN EAST AFRICA

(After Burton, 1979)



The diagram does not attempt to show all the animals involved in the savanna pyramid, but illustrates representatives of the various groups.

1. EFFECT OF SOIL DEPTH, TEXTURE, pH AND WATER CONTENT ON THE DISTRIBUTION OF ORGANISMS

(A, B; II)

Objectives

1. to understand the relationship between various soil conditions and distribution of organisms.
2. to relate soil texture and structure to the distribution of organisms.
3. to relate soil water content to the distribution of organisms.

Curriculum links

Conservation, soil composition, soil organisms.

Preparation

Teacher: Identify and visit the area where samples are to be taken. Assemble all the required materials and equipment. Divide the students into groups of 3 or 4, each group assigned to a particular area.

Students: Knowledge of classification of soils on the basis of structure and texture. Skills of sampling and identification using dichotomous keys. Skills of determining soil water content and pH.

Equipment: See Chapter Four, page 36 'Soil Analysis' etc.

Strategy

Question: To what extent do water content and pH relate to the distribution of organisms in the soil?

1. Obtain soil samples from different areas in a savanna ecosystem e.g. open grass, under a trees and bare soil.
2. Identify the organisms found in each sample. Count the number of species in the sample.
3. Classify the samples on the basis of soil structure.
4. Determine the water content of each sample by drying the samples to constant weight.
5. Determine the pHof each sample.
6. Relate the distribution of each species to the above factors.

2. EFFECT OF COVER ON THE DISTRIBUTION OF ANIMAL AND PLANT AND SPECIES

(A, B; II)

Objectives

1. to understand the relationship between cover and distribution of animal and plant species.
2. to acquire the skill of sampling plant and animal species along a line transect.

Curriculum links

Species diversity, light and plant distribution, competition.

Preparation

Teacher: Identify and visit the area where study is to be carried out. Obtain permission if necessary. Assemble all the required materials and equipment. Divide the students into working groups of 4 or 5.

Students: Knowledge of fauna and flora and factors affecting their distribution. Skills of identification and classification using a dichotomous key.

Equipment: See Chapter Four, page 14.

Strategy

Question: To what extent does cover determine the distribution of animals and plants?

Each Group -

1. Lay a line transect in a wooded area within the savannah grassland.
2. Determine the quadrat size to be used for sampling stations.
3. Determine the unit interval between sampling stations.
4. Count the number of each species in each quadrat.
5. Record the presence or absence of cover for each quadrat.
6. Relate the effect of cover to the distribution of animal and plant species.
7. Students to compile a list of plant and animal species found in shade and another list of those found in the open.

Follow up

Have the students discuss whether or not the distribution could be related to other factors apart from cover.

3. TO DETERMINE THE DOMINANT SPECIES OF PLANTS AND ANIMALS IN AN OPEN GRASSLAND

(A, B, D; II)

Objectives

1. to identify the dominant species of plants and animals in an open grasslands.

2. to acquire skills of sampling and identification.

Curriculum links

Plant and animal diversity, concept of dominance.

Preparation

Teacher: Identify the area to be used for the study and, if necessary, ask permission to use it. Assemble all the necessary materials and equipment. Divide the students into groups of 5 or 6, each to work on a different area within the same open grassland.

Students: Knowledge of grassland fauna and flora. Skills of identification and classification. Skills of sampling using a quadrat.

Equipment: See Chapter Four, page 14.

Strategy

Question: How are dominant animals and plants distributed?

Each Group -

1. Prepare an outline map of whole area and mark the presence of very large plants e.g. trees and animals e.g. antelope. Note movements of large animals.

2. Obtain samples of the open grassland by throwing quadrat randomly.

3. Count and record the number of each species of plants and animals in each quadrat.

4. Obtain samples in this way 5 or 6 times, each time counting the number of each species.

5. Tabulate the results of the species count from all the quadrats.

6. Determine the dominant species of animals and plants in the open grassland in terms of size and numbers.

7. Discuss the idea of dominance and the ecological effects of dominant organisms.

Follow up

Have the students relate the effect of dominance to various ecological conditions.

4. TO COMPILE DESCRIPTIONS OF ECOSYSTEMS

(A, B, C; I,II)

(This activity is linked to Activities 1 and 3)

Objectives

1. to describe the inter-relationships of organisms in savanna ecosystems.
2. to identify and construct food chains, food webs and pyramids of numbers in savanna ecosystems.

Curiculum links

Diversity, ecosystem, food chains and food webs.

Preparation

Teacher: During activities 1 and 3 get one or two students per group to note down any observations on the feeding habits of the animals.

Provide reading for the students on the feeding habits and diet of animals in savanna.

Strategy

Question: How are animals and plants in a habitat related to one another?

1. Have students compile tables one for the soil and another for the open grassland habitat. The tables should list the animal species, numbers, and the types of food eaten by each species.

2. Using the above data help students to construct

   (a) a food chain

   (b) food web

   (c) a pyramid of numbers, for each habitat.

3. With reference to these discuss the concept of "balance of nature".

Follow up

1. Discuss ways in which man could interfere with the balance of nature.

2. Have the students carry-out similar activities in different seasons (wet and dry). This will enable them to establish whether the organisms in those habitats are permanent dwellers or seasonal. Get them to attempt to explain any changes which might have taken place.

5. EFFECT OF GRASS COVER ON THE DISTRIBUTION OF ANIMAL POPULATION

(A, B, C; d; I, II, III)

Objectives

1. to compare the distribution of animal populations as related to cover.
2. to acquire the skill of experimenting with equal plots and singling out experimental factors.
3. to appreciate some aspects of the management of grasslands.
4. to appreciate the effect of overgrazing on the distribution of some animals.

Curriculum links

Animal distribution, experimentation, grassland management.

Preparation

Teacher: Visit the study area and obtain the necessary permission for the visit. Divide the students into working groups of not more than 6.

Students: Knowledge of fauna derived from Activities 1 to 4. Skills of measuring, identification and counting.

Equipment: large scissors
pegs and line
1m rule

Strategy

Question: How does amount of grass effect distribution of animals?

1. Select two $1m^2$ plots of similar grassland.
2. Have the students estimate the types and numbers of animals in each plot, allowing for movement in and out of the plots.
3. With scissors cut off all the vegetation cover at soil height and remove from one of the plots while leaving the other with the vegetation undisturbed.
4. Leave the two plots undisturbed for a period of 2 days. Water them equally and observe, from time to time, any movement of animals (small and large).
5. After the 2 days estimate the number of various animal types in each plot.
6. Account for any differences.

Follow up

Have the students compare some non-grazed with over-grazed areas. Discuss problems concerned with management of grasslands.

6. EFFECT OF THE AMOUNT OF GRASS COVER ON THE DISTRIBUTION OF ANIMAL POPULATIONS

(A, B, C; d; I, II, III)

This is a more elaborate version of Activity 5.

Objectives

1. to acquire the skills of experimenting with equal plots and singling out experimental factors.
2. to realise the effect of non-grazing, undergrazing and over-grazing on the distrubition of animals.
3. to appreciate factors affecting the adequate management of grasslands.

Curriculum links

Distribution of animals, conservation and use of grasslands.

Preparation

Teacher: Visit the area and obtain any necessary permission for the visit. Divide the students into working groups of not more than six. Prepare equipment for measuring and cutting. (See Activity 5)

Students: Knowledge of fauna derived from previous activities. Skills of identification, counting and measuring.

Strategy

Question: Does the height of grass influence the distribution of animals?

1. Select 4 plots of similar grassland each of $1m^2$.
2. Have the students estimate the numbers of the various types of animals within the plots.
3. From one of the plots remove all the vegetation cover.
4. From the second plot, cut the grass to a quarter of the original height.
5. From the third plot, cut the grass to a half of the original height.
6. From the fourth plot, cut the grass to three quarters of the original height.
7. Water all the four plots equally for a period of 2 days.

8. After the two days, go back and estimate the numbers of the various animals types in each plot. (If possible also make observations from time to time on any animal movement.)

9. Account for any differences observed, referring also to the control plot (uncut) in Activity 5.

Follow up

1. Investigate whether the cutting of the grass affected the distribution of other plants in the plots.

2. Discuss the implications of the findings of this investigation for grassland management.

7. EFFECT OF TILLING ON THE DISTRIBUTION OF ANIMALS IN THE SAVANNA ECOSYSTEM

Objectives

1. to explain the relationship between tilling and the distribution of animals in the savanna ecosystem.
2. to acquire the skill of experimenting with equal plots and singling out experimental factors.
3. to appreciate the effects of tilling on the distribution of animals.
4. to appreciate adequate management of grasslands.

Curriculum links

Distribution of animals and plants, conservation and use of grasslands.

Preparation

Teacher: Visit the area and obtain any necessary permission. Divide the students into groups of not more than six. Prepare equipment for measuring (line, rule and pegs) and digging (e.g. spades).

Students: Knowledge gained from Activities 1 - 6. Skills of identification, counting and measuring.

Strategy

Question: How does digging effect the distribution of plants and animals?

1. Select two plots of similar grassland of $1m^2$ each.
2. Have the students dig up one of the plots.
3. Leave the other plot undisturbed.
4. Water the two plots equally and leave them undisturbed for two days.
5. After the two days, make a count of the animals and plants and their types in each plot.
6. Discuss possible reasons for the differences.

Follow up

1. Discuss why **all** the plots used in Activities 5 - 7 were watered. What might have happened if they had not been? Carry out investigations to test hypotheses.
2. Have the students make observations on tilled and untilled grasslands on farms.
3. Relate the results of this investigation to farming methods.

8. CHANGES IN FAUNA AND FLORA IN THE SAVANNA ECOSYSTEM

(A, C; d; II)

Objectives

1. to understand the relationship between seasons and changes in biotic communities.
2. to acquire skills of analysing data.

Curriculum links

Change of biotic communities with time, conservation and use of grasslands.

Preparation

Teacher: Obtain data on the variations of animal and plant communities over a period of one year from a National Park headquarters or similar body.

Students: Knowledge of main fauna and flora in natural parks. Understand concepts of food chain, food web and pyramid of numbers. Skills of analysing data.

Strategy

Question: How is the distribution of animals and plants affected by the seasons?

1. Supply the obtained data to the class.
2. Have the students plot a graph of the numbers of the respective animals and plants over the 12 months of a year.
3. Assign the animal communities to trophic levels.
4. Relate the fluctuations of the number of animals and plants with the weather patterns.
5. Relate the weather patterns to the availability of food at different trophic levels.
6. Discuss possible reasons for the fluctuations of the various plant and animal communities.
7. Using the data obtained construct food chains, food webs and pyramids of numbers for the ecosystem.

Follow up

Have the students investigate the fluctuation of biotic communities over various seasons in grassland areas near to their school.

9. SECONDARY SUCCESSION IN A GRASSLAND ECOSYSTEM

(B, C; a; II, III)

Objectives

1. to identify the series of biotic communities that characterise secondary succession.
2. to compare the ecological relationships of biotic communities in secondary succession.
3. to acquire the skill of experimenting and analysing data.
4. to appreciate factors affecting adequate management of grasslands.

Curriculum links

Ecological succession, conservation and use of grasslands.

Preparation

Teacher: Visit the study area and obtain any necessary permission. Divide the class into working groups of not more than 6.

Students: Background knowledge of fauna and flora.

Strategy

Question: Is there a pattern in the way bare land is colonised?

1. Select two plots of $1m^2$ each of similar grasslands.
2. Have the students burn the grass in one of the plots.
3. Leave the other plot with its grass cover.
4. Water the two plots similarly and observe and record the fauna and flora that colonises the burnt area at monthly periods.
5. Discuss any change in biotic communities with time.
6. Compare the monthly observations with observations on the control plot.
7. Produce a detailed graph showing the pattern of succession over time.

NB: This is an investigation which can run for a year or more.

Follow up

Discuss the implications of the results of this investigation for the control of slash and burn methods of farming.

10. THE EFFECT OF LIGHT, MOISUTE AND HEAT ON THE DISTRIBUTION OF TERMITES

(A, D; I, II, III)

Objectives

1. to describe the behaviour pattern of termites with respect to light, moisture and heat.
2. to attempt an explanation for the kind of behaviour observed.
3. to relate the type of behaviour and its significance to the natural habitat of the animals.
4. to formulate possible control experiments to investigate this behaviour.

Curriculum links

Adaptation, behaviour, taxes and kineses.

Preparation

Teacher: Take class to see one or two termite nests. With care expose part of the internal structure of a nest and get students to make notes (or drawings) of its appearance. Collect samples of termites from the nests for investigations to be carried out in the school.

Students: Obtain background knowledge on the habitats of termites and their economic importance.

Equipment: Per group of not more than six students -

3 or more Petri dishes
black covering e.g. muslin cloth
water absorbent material e.g. paper, cloth, cotton wool

Strategy

Question: What factors influence the distribution of termites in a termite hill?

1. Each group of students prepare one or more Petri dishes so that one half is dark and the other exposed to light as shown below.

Light Dark

2. Place 20 termites in the light half and 20 termites in the dark half of each Petri dish. Leave for an hour.

3. After one hour count the number of termites in each half of the Petri dish.

4. Discuss the results of the investigation. How might they relate to the distribution of termites in the natural habitat?

5. Discuss methods of control for this investigation. (For example, all termites put in one end at start.)

6. Each group of students prepare one or more Petri dishes with the walls of one half lined with wet filter paper while the other half is lined with dry filter paper. The top and bottom are not lined.

7. Place 20 termites in the damp half and 20 in the dry half of the dishes.

8. Count the number of termites in each half after about 30 minutes.

9. Discuss the results of the investigation. How might they relate to the distribution of termites in their natural habitat?

10. Discuss methods of control in such an investigation.

11. Repeat 6 - 10 but instead of lining the Petri dishes with filter paper wrap one half in damp cottom wool and the other half in dry cotton wool. At step 8 of the investigation take the temperature of the two halves of the Petri dish besides counting the termites in them.

Follow up

1. The three investigations are concerned with the effect of light, humidity and temperature on the behaviour of the termites. Get students to devise and carry out an investigation to determine what combination of these factors has the greatest effect.

2. Undertake investigations to determine whether it is speed of movement (kinesis) or direction of movement (taxis) of the termites which produced the results obtained.

3. Repeat the investigation using land snails or slugs. Larger glass vessels than Petri dishes are more convenient for this.

11. AN INVESTIGATION OF THE BEHAVIOUR OF LAND SNAILS WITH RESPECT TO A HORIZONTAL AND VERTICAL COLUMN

(D; II, III)

Objectives

1. to describe the behaviour pattern of snails with respect to the vertical and horizontal orientation of a container.
2. to relate such behaviour to their life in their natural habitat.

Curriculum links

Orientation behaviour, ecological significance of orientation behaviour.

Preparation

Teacher: Provide each group of students with a bucket or similar vessel with vertical sides of about 25 cm and a diameter of about 15 cm. Collect 3 - 5 land snails for each group or get students to do so.

Strategy

Question: How do snails distribute themselves on vertical and horizontal surfaces?

1. Introduce the snails into the bucket and let them settle at the bottom.
2. Place the bucket vertically by a window and observe the behaviour of the snails.
3. After a period of at least half an hour place the bucket on a table horizontally.
4. Again observe the behaviour of the snails in the bucket.
5. Discuss any differences noticed about the behaviour of the animals when the bucket was vertically placed and when it was horizontally placed.
6. Obtain explanations for this kind of behaviour. Discuss how such behaviour relates to the snails' habitat.
7. Get students to suggest possible control experiments which could confirm their observations and explanations.

12. AN INVESTIGATION OF THE BEHAVIOUR OF BIRDS RELATING TO A FEEDING TABLE

(D; I, II)

Objectives

1. to describe the behaviour of birds when they come to feed on a feeding table.
2. to identify birds visiting the feeding table when different kinds of food are placed there.
3. to attempt explanations for the kind of behaviour pattern they will have observed.

Curriculum links

Feeding behaviour, animal mouth parts.

Preparation

Teacher: Consult Chapter 5 'Ethological Methods' (page 58) in order to devise suitable observation procedures.

Students: Revise feeding habits of birds.

Equipment: a feeding table (a piece of wood or similar firm material (say 0.5 m^2) fixed on a pole (say 1.5 m high) or to a tree or other vertical structure
pieces of meat
pieces of cooked maize-meat (Ugali)
maize grains or any other grains
a pair of binoculars (optional)

Strategy

1. Construct a feeding table in open ground in a grassland savanna area.

2. Place different foods (meat, maize meal, grains) on the feeding table.

3. Arrange students in groups of 3 or 4 to prepare observation schedule to answer the following or similar questions.

   What types of birds come to the feeding table?

   How do the birds approach the table?

   Do the birds arrive singly or in groups?

   Do you, at any time, find more than one bird type on the table?

   Which types of birds eat which types of food?

   Did you notice any fighting among birds of different species? If there was, what explanation would you give for it?

What happens if you chase the birds away from the table but then quickly retreated to watch them from a distance? Do all of them or some come back to the feeding table or does another lot come in their place?

5. Get students to draw hypotheses about the birds feeding behaviour and devise experiments to test them e.g. placing different kinds of food on the table to determine preference by different species.

6. Discuss the relevance of the birds' feeding habits to the ecology of the area in which they are found.

Follow up

1. Carry out the experiments students have devised.

2. Discuss the relevance of studies of animal behaviour to ecology, especially in evaluating the significance of animals in a food web.

13. EFFECT OF GRAZING ON THE GROWTH OF GRASS IN SAVANNA

(B; b; II, III, IV)

Objectives

1. to understand the influence of factors affecting the growth of grass in savanna.
2. to determine the influence of grazing on the rate of growth of grass.
3. to acquire the skill of experimenting with equal plots and singling out experimental factors.
4. to realise the effect of overgrazing and undergrazing on the growth of grass.
5. to appreciate the importance of adequate management of grassland.

Curriculum links

Plant growth, conservation, grassland management.

Preparation

Teacher: Visit an area of grassland before the students in order to ascertain the most suitable plots for experiments. If possible use plots in the school grounds. Divide students in working groups of not more than 5 - 6 each. Each group to be responsible for a plot of grass.

Students: Revise knowledge of grassland in savanna and ecological factors affecting them. Revise skills of measuring and weighing.

Equipment: string and small stakes
scissors

Strategy

1. Select 3 plots of similar grassland of about 1 m^2 each. (Use multiples of 3 plots for more students.)
2. Mark out the plots using string and stakes.
3. Have the students write out the characteristics e.g. species and physical factors, of each of the plots.
4. With scissors cut the grass of one plot to a height of 2 cm. Weigh the cut grass.
5. Cut the third right down to the soil surfaces. Weigh the cut grass.
6. Leave one plot uncut - as a control.
7. Water the three plots equally and leave them for two weeks.
8. After two weeks, cut the grass in all of the plots to a height of 2 cm. Weigh the amount of grass cut from each plot.
9. Which plot has the most harvest?
10. Get students to account for the differences in harvests.

Follow up

1. Calculate the aggregate harvests from the plots i.e weight of grass cut initially plus weight of grass cut after two weeks. Discuss the results.
2. Get students to formulate hypotheses about the relationship between grazing and growth of grass.
3. Have the students observe different areas of savanna grassland and compare non-grazed areas with well-grazed and over-grazed areas, and relate their observations to their experiments.

14. THE EFFECT OF BROWSING ON THE GROWTH OF GRASS IN THE SAVANNA

(B; b; II, III, IV)

Objectives

1. to understand the relationship between some factors affecting the growth of grass in the savanna.
2. to determine the rate of growth of grass as related to nipping off (browsing).
3. to acquire the skill of experimenting with equal specimens and singling out experimental factors.
4. to appreciate the importance of adequate management of grassland.

Curriculum links

Plant growth, conservation, management of grasslands.

Preparation

Teacher: Make a reconnaisance trip to the area and get permission from relevant authorities. Divide students in working groups of not more than 3 each.

Students: Revise knowledge of flora and browsing animals in savanna. Revise skills of measuring and recording.

Equipment: netting to cover 1 m^2 and 40 m x 30 cm.

Strategy

1. Fence off a small plot (1m^2) with netting. Also cover with netting.
2. Have the students identify several tufts of grass of equal height.
3. Have the students record their height.
4. Have the students nip off the tips of half of the grass tufts. Measure and record the average length nipped off.
5. Leave the other tufts untouched.
6. Repeat the nipping off every week on the same tufts and keep a record of the average length nipped off.
7. Water the plots equally and carry on the nipping off for several weeks.
8. After a period of not less than one month students should add average height measurements of the tufts harvested by nipping off to their original height.

9. Then they measure the height of the grass tufts that were left intact and calculate the average.

10. Determine which tufts had greatest growth and account for the difference.

11. Discuss the implications of this experiment for grassland and management.

Follow up

Have students observe different areas of savanna grasslands and compare those that have not been exploited by browsers, those which are browsed and those over-browsed. Get them to devise experiments to test any hypotheses drawn from their observations.

15. THE EFFECT OF BURNING AND DIGGING ON THE GROWTH OF GRASS

(B; b; II, III, IV)

Objectives

1. to understand the effect of burning on grass growth.
2. to observe plant succession.
3. to acquire the skills of experimenting with equal plots and controlling variables.
4. to appreciate the importance of adequate management of grassland.

Curriculum links

Plant growth, conservation, grassland management.

Preparation

Teacher: Obtain a suitable area of grassland near to the school and where burning will not be dangerous. Divide the students into groups of not more than 6 in each.

Students: Revise knowledge of flora of savanna. Revise skills of measuring.

Equipment: string (4m) and pegs (4) for four plots of 4 x 1m per student group
scissors

Strategy

1. Mark out four 1m plots.
2. Draw vegetation maps of each plot.
3. Burn the grass on one plot. Cut the grass to ground level on another plot. Dig up and remove grass from another plot. Leave the fourth plot untouched as control.
4. Water the plots regularly and equally.
5. Over a period of a month observe the plots regularly and note any changes in the vegetation.
6. Construct a vegetation map of each plot at the end of the month.
7. If possible repeat these procedures over several months.
8. Compare the vegetation maps for each plot and determine the effects of burning and digging.
9. Discuss the concept of plant succession as illustrated by the experiment.

Follow up

Discuss the advantages and disadvantages of controlled burning in grassland management.

16. THE EFFECT OF CONTROLLED BURNING ON SOIL PRODUCTIVITY

(B; b; II, III, IV)

Objectives

1. to understand the relationship between some factors favouring the growth of grass in the savanna.
2. compare the productivity of soil as related to burning.
3. to acquire the skill of experimenting with equal plots and singling out experimental factors.
4. to appreciate the effect of burning on the productivity of soil.
5. to appreciate some of the issues concerned with farming in grasslands.

Curiculum links

Plant growth, conservation.

Preparation

Teacher: Obtain a suitable grassland area for the work near to the school. Obtain bean or comparable seeds. Divide students into working groups of not more than 6 each.

Students: Revise knowledge of flora in savanna. Revise skills of measuring and weighing.

Equipment: hoe
simple balance for weighing
matches

Strategy

1. Select 2 plots of similar grassland of about $1m^2$ each.
2. Using a hoe, dig up the grass and remove from one plot.
3. Burn the grass in the other plot, then dig up the plot.
4. Plant an equal number of good quality beans in both plots in equal rows of equal spacing.
5. Water and weed the plots equally and leave them for 4 weeks.
6. After four weeks harvest all the bean plants.
7. Calculate total weight of harvest for each plot.
8. Determine which plot has the greatest harvest. Discuss possible reasons (hypotheses) for any differences.
9. Based on the results of this investigation and those of others done previously get students to draw up a policy for adequate grassland management.

Follow up

Discuss the advantages and disadvantages of slash and burn methods of agriculture. Have the students undertake projects to test the hypotheses of reasons for different effects of burning and hoeing.

DESERTS

ARID AND SEMI-ARID LANDS

DR. M. S. SELIM

DESERTS - ARID AND SEMI ARID LANDS

INTRODUCTION

Deserts are arid zones. They occur in regions with less than 10 inches of annual rainfall, or sometimes in hot regions where there is more rainfall but it is unevenly distributed in the annual cycle. In addition through his misuse of grasslands and forests by, for example, uncontrolled slash-and-burn methods of agriculture or allowing over-grazing, man has produced deserts.

Four very distinctive life forms of plants are adapted to the desert ecosystem: annuals which avoid drought by growing only when there is adequate moisture; desert shrubs with numerous branches arising from a short basal trunk and small, thick leaves that may be shed during the dry season when the plant becomes dormant before wilting occurs; the succulents which store water in their tissues; and the microflora such as mosses and blue green algae that remain dormant in the soil but are able to respond quickly to cool or wet periods.

Seeds of many annual desert plants sprout only when there is rain of a certain minimum magnitude (12mm of rain or more). The mechanism here seems to involve a chemical germination inhibitor that must be washed off the seed coat. Annual desert plants grow, bloom, fertilise and produce seeds, all within a matter of days after rain, so as to avoid the dry long season. They are drought evaders.

Desert shrubs are widely spaced. Sometimes they look uniformly distributed as if planted at regularly spaced intervals. The pattern can be explained by competition for scarce water which eliminates all but one individual in a given area. Evolution of self-regulation of the population has occured in some species. The severe competition for water is avoided by the production of leaf or root hormones that inhihit the development of other plants in the neighbourhood. Chemical substances released by decaying leaves that fall to the ground under the shrub or by the living roots in the soil inhibit or kill any seedling that may start to sprout. Such a control mechanism tends to keep plants spaced apart, thereby reducing actual competition for water.

Animals such as reptiles and insects are adapted to deserts. Their impervious integuments and dry excretions enable them so survive on the small amount of water produced in the body as a result of the breakdown of carbohydrates. Mammals as a group are poorly adapted to deserts, but a few species have become adapted. Some nocturnal rodents, for example, that excrete very concentrated urine and do not use water for temperature regulation, can live in the deserts without drinking water. Other animals such as camels must drink, but are able to survive without drinking for several days.

Semi-Arid Zones

Where water is a limiting factor, but the quality of the soil is good, irrigation can convert deserts into productive agricultural land. Many such semi-arid zones are productive areas. Whether productivity continues or not, depends on how well man is able to stabilise biogeochemical cycles and energy flow at increased rates. As the large volume of water passes through the irrigation system, salts may be left behind that will gradually accumulate over the years until their poisonous effects become a limiting

factor. The water supply itself can fail if the watershed from which it comes is abused. The existence of productive semi-arid zones depends on such difficulties being overcome and this depends on careful management based on ecological principles.

1. MAPPING AN ARID HABITAT

(A, B, C; I, II)

Objectives

1. to develop an understanding of the physical factors affecting organisms in arid habitats.
2. to identify plants and animals in the area and understand the interrelations among organisms.
3. to understand the unity and diversity of living organisms and the complexity of arid ecosystems.
4. to understand the structure and function relationship among animals and plants in arid habitats.
5. to develop skills in using field equipment.
6. to acquire positive attitudes and values towards the environment.

Curriculum links

Variety of life, classification, adaptation.

Preparation

Habitat: near to the school. Check that organisms are present.

Equipment: map including area to be studied
tape mesures
simple identification keys
insect-catching equipment (see page 14)
thermometers

Strategy

Question: What kinds of animals and plants are found in the habitat and how are they adapted to their environment?

1. Map the area: If the area is large, trace an outline from an appropriate map. If a small area is chosen use a tape measure to produce a scaled down outline map. Mark important geological and other features on the map.

2. Record essential data. The following information should be recorded: date of visit, location of habitat, annual rainfall.

3. Plants of the habitat:

   (a) Try to identify the plants in the habitat, using a key. This will avoid unnecessary collecting.
   Remember plants are often scarce in arid conditions

   If you have to take a flowering plant away for identification, remember to include some leaves of the plant too. When listing the plants use the following titles: Date, Common name, Latin Name, Family, Stage of Growth, Average height.

   (b) Investigate and record the frequency of plants in an identified small area (10 x 10 metres). Encourage students to think of reasons for the distribution of plants.

   (c) Record how frequently plants occur using the following symbols:

   d - dominant. This would be used for the species which either because of its size or numbers, would if removed bring about a great change in the appearance of the habitat.

   a - abundant. Found in large numbers.

   f - frequent.

   o - occasional.

   r - rare.

   vr - very rare.

   (d) If you notice that other areas have species of plants which appear to differ in size or number from those in the area under investigation, try to find reasons for this. Is it possibly due to:

   - sudden change in soil types?
   - trampling by mammals?
   - exposure to greater amounts of light?
   - steep slopes?

4. Animals of the Habitat:

   (a) Collect some insects and place them in specimen tubes. Use a sweepnet and hold a white sheeting under the bushes to catch insects when the vegetation is shaken. Try to identify the insects.

   (b) List the birds seen. Why are they few? Are they resting? Are they feeding? Are they building nests?

(c) Look for lizards and other reptiles near the vegetation.
Be careful of snakes. Never put your hand in a hole.

(d) List the mammals seen. What are the most abundant mammals?

(e) Why do arid areas seem empty of animals during the heat of the day but become alive towards evening?

5. The Temperature of the Habitat:

Make records of the temperature at different levels in the air, under leaf litter and at soil level. Do this at intervals of two hours during the day. Draw graphs to show:

(a) The relation between temperature and time intervals at soil level.

(b) The relation between temperature and time intervals under leaf litter.

6. Laboratory Work:

(a) Identify the organisms collected using the available flora, and fauna keys.

(b) Investigate the morphology (form and structure) of the organisms and draw diagrams representing them.

(c) Work out how the prevailing conditions of the arid zones have their impact on the morphological features.

2. PLANT DISTRIBUTION: RICH HABITAT, DRY SEASON

(A, B, C; a; I, II)

Objectives

In addition to objectives of Activity 1.

1. to investigate and record the frequency of plants in an area of a rich habitat.
2. to appreciate the relationship of physical factors, especially humidity, on plant distribution.
3. develop the ability to draw plants and the skills of reading diagrams.

Curriculum links

Adaptation, classification, distribution.

Preparation

Habitat: A rich habitat in an arid zone may be located around a well, spring, or near the bank of a lake or river passing through an arid area. Identify the location before taking students to perform the activity.

Equipment:
measuring tape
poles
string
hand lens
forceps
plastic bag
scale
burner
crucible
holder

Strategy

Question: To what extent does humidity influence the distribution of plants?

1. Choose suitable plots and mark them by the string and poles. (Divide students into small groups and each group investigates a plot for comparison.)
2. Record the following data: date, time, location, annual rainfall and source of water.
3. Identify plants by using a key.
4. Give different symbols for each plant (x, ≠≠, *, ... etc.) and make a diagrammatic representation of its distribution in the plot.
5. Determine the frequency of occurance (d, a, f, o, r, or vr).
6. Determine the amount of humidity in soil samples. (See page 36)
7. Compare the frequency of plants in the different plots and attempt to explain any differences.

Follow up

1. Take samples of plants.
2. Identify the plants and make drawings of them.
3. Make an exhibit showing the distribution of plants in a rich arid zone habitat using natural samples and a diagram.
4. Provide explanations for the nature and frequency of plants existing in such a habitat. Why are many plants succulent?

5. Undertake a parallel investigation using the same preparation and strategies as in this Activity but based on a 'poor' habitat located away from any water source.

   Compare the types and distribution of plants in the 'poor' and 'rich' habitats. Work out reasons for any differences bearing in mind that both investigations are taking place in the dry season.

3. PLANT DISTRIBUTION: RICH HABITAT, RAINY SEASON

(A, B, C; a; I, II, IV)

Objectives

Besides objectives mentioned in Activities 1 and 2.

1. to observe the change in plant distribution and the stages of development affected by increase of humidity (rain).
2. to work out reasons for the change in the habitat.
3. to appreciate the relationship of water to plant life in arid zones.
4. to develop scientific skills and attitudes.

Curriculum links

Adaptation, classification, sampling.

Preparation

Equipment: As in Activity 2.

Watch for rain and prepare your students for a second visit to the plots used in Activity 2. Make your visit between ten days and two weeks after rain. Have your students take their notes of Activity 2 for comparison. It is advisable that each group study the same plot they studied in the previous activity.

Strategy

Question: What is the impact of rain on the plant life of arid areas?

1. Have your students go through the steps of Activity 2.

2. Make comparisons based on the following -

   (a) Changes in plant distribution

   (b) The occurance of new plants not seen before.

(c) Any morphological changes besides the size of the plants: abundance of leaves, flowers ... etc.

Follow up

1. Collect plant samples and make drawings to show their distribution and frequency, including new plants if any.

2. Make an exhibit to compare with that of Activity 2 to show the changes.

3. Determine whether the distributions found are similar to those of other parts of the desert.

4. Undertake a parallel investigation using the same preparation and strategies as in this activity but using a 'poor' habitat located away from a water source.

   Compare the types, adaptations and distribution of plants in the 'poor' and 'rich' habitats following rain. Work out reasons for any differences bearing in mind that water is now available in both habitats and that factors such as soil type and seed distribution may be influential.

4. ANIMAL DISTRIBUTION IN AN ARID HABITAT

(A, D; I, II)

Objectives

1. to identify animals in an arid habitat.
2. to comprehend the unity and diversity among animals in an arid zone ecosystem.
3. to develop skills of mapping animals.
4. to understand the effect of abiotic (physical) factors on animal distribution in arid habitats.
5. to acquire positive attitudes towards the environment.

Curriculum links

Insects, reptiles, birds, mammals, classification, adaptation.

Preparation

Teacher: Since animals are not abundant in arid zones specific directions should be given to the students to help in observing the animals they are likely to observe. This requires surveying large areas, being quiet, and making careful observation. (See Chapter Five, page 58) Divide students into groups of five or six.

Students: Revise major anatomical and physiological features of animals likely to be observed.

Equipment: nets traps and jars for collecting specimens (See Chapter Four, page 14)
magnifying glass
binoculars (if possible)

Strategy

Question: How are animals distributed in arid conditions?

1. Each group of students should make arrangements for observing animals and their behaviour in a different area by reference to the principles and techniques outlined in Chapter Five. Note that observations should include looking for evidence of the presence of animals e.g. footmarks and browsing marks on plants, as well as observing them directly.

2. Using a simple key identify the animals observed.

3. Construct a sketch map of the distribution of animals in the area including indications of any movements e.g. flights of birds, burrowing of lizards in sand, grazing movements of mammals.

4. Construct a sketch map of the same area noting its main physical features e.g. type of soil cover, plant cover, temperatures, light and shade and presence of water.

5. Draw up and discuss conclusions (hypotheses) relating the distribution of the animals to their behaviour and their environment.

6. Using information gained from stages 1 and 3 and ideas from Chapter Four attempt to catch one specimen of each animal species. Do <u>not</u> catch species that are few in number and make certain the animals remain alive.

Follow up

1. Compare the distribution of animals in habitats with different environmental features e.g. rocky v sandy and oasis v dry desert.

2. Have the students construct a model of the habitat they have studied using materials e.g. sand, pebbles, plant material, gathered from the habitat and arranged on the floor or a bench in a classroom. Arrange animals in containers providing them with food and suitable physical conditions on the model to demonstrate their distribution. Such an activity can help students synthesise knowledge they have gained from specific studies.

5. ORGANISMS IN ARID SOILS

(A, B; I, II, III)

Objectives

1. to identify organisms prevailing in soil types of arid zones.
2. to understand the interrelationships or organisms in soil and arid zones ecosystems.
3. to acquire the skills of investigation, identification, and classification of soil organisms.

Curriculum links

Adaptation, classification, soil ecosystems.

Preparation

Teacher: Refer to Chapter Four.

Equipment:
microscope
magnifying lens
light
filter funnel
muslin cloth
flasks
lime water

Strategy

Question: How are different organisms distributed in arid soils?

1. Obtain samples of arid zone soil from different locations and depths. Crumble up the samples in dishes.

2. Observe and identify organisms that can be seen by the naked eye or by a magnifying glass.

3. Put each sample in a muslin cloth and hang in a flat bottom flask with a little clear lime water in the bottom. Close the flask and leave overnight. Examine the lime water before and after. Discuss the findings (a white precipitate indicates the presence of respiring organisms).

4. Place a layer of cloth in each filter funnel and place in it a soil sample.

5. Place the funnel in a conical flask or a beaker containing water. Make sure the wall of the flask touches the tube of the funnel.

6. Expose the top of the funnel to the heat and light of a strong light bulb and leave for sometime. (See Figures 14, 15 and 16 page 53, Chapter Four for details of this method.)

7. Observe organisms in the water using a hand lens and a microscope.

8. Compare with other samples of soils, testing for organisms in the same way.

9. Tabulate the results and discuss - What kinds of soil has most organisms in them? Why? Is there any relationship between presence of certain organisms at different depths of soil? Why?

6. STUDYING A HABITAT AT NIGHT

(A, C; I, II)

Objectives

1. to develop a distinct concept of desert life at night.
2. to appreciate the dynamics in an ecosystem between day and night.
3. to observe the changing relationship of living organisms at night.
4. to investigate the extreme changes in certain climatic conditions between day and night.

One of the major features of deserts is the considerable relative contrast in temperature and in some areas, humidity, between night and day. An understanding of the differences between night and day (the diurnal cycle) is important for ecology in desert regions.

Curriculum links

Diurnal cycles.

Preparation

Teacher: Use the same location used for a previous activity during the day. Make a preliminary visit at night to ensure there are no features which might cause accidents or make observations difficult.

Establish clear procedures for students so that they remain in contact with each other and the teacher.

Ensure students have warm, protective clothing. Arrange students in groups of 5 or six.

Students: Review the differences between conducting field work at night and in the day. Practice using observation schedules.

Equipment: As previous activities plus lamps. See also Chapter Four for methods of measuring temperature and humidity.

Strategy

Question: In what ways is a habitat different at night?

1. Arrange observation locations of students in each group. Make as many observations without a lamp as possible. (Our eyes get used to the dark after a while.)

   At first concentrate on observations of animals and record them.

2. Make observations of animals using quick flashes of lamps.

3. Light the area as fully as possible and record any further observations on animals, especially ones under plants.

4. Make a sketch map of the plant cover and note any features different from those of the day.

5. Take samples of soil temperature in different locations and at different depths.

6. Take samples of soil for measuring humidity content.

7. Compare maps of distribution of animals and plants from this activity with those prepared in the day time.

8. Relate differences in distribution and characteristics of organisms to differences in the physical environment between day and night.

Follow up

At an opportune moment compare a habitat at night before and after rainfall.

7. A STUDY OF AN OASIS (OR IRRIGATED AREAS)

(A, B; a, d: I, II)

Whilst arid regions typically have very low rain falls some (semi-arid) areas in them have a continuous source of water which allows cultivation to be undertaken. The water supply might be underground and appear in the form of springs or wells. Rivers can be another source of water.

While plants and animals in arid zones are mainly wild, in such semi-arid areas a mixture of arid flora and fauna with other cultivated and introduced plants and animals is to be found.

Oasis are green areas surrounded by sandy, yellow desert, where springs and wells are the main source of water. This under-ground water enables wild life to grow and attracts human beings to settle and cultivate the land.

Objectives

1. to formulate the concept of an oasis in an arid area.
2. to appreciate the main factors contributing to the formation of an oasis.
3. to understand the abiotic (physical) factors affecting living organisms in an oasis.
4. to identify the plants and animals in an oasis and understand the interrelationships between organisms and between biotic and abiotic factors.
5. to appreciate the complexity of the oasis ecosystem.
6. to investigate human settlements and activities in an oasis.
7. understand the various climatic conditions prevailing in different seasons of the year.

Curriculum links

Ecosystems, agriculture, land management.

Preparation

Teacher: Undertake a preliminary survey of the oasis or comparable irrigated area. Assign students to groups of 5 - 6.

Students: Revise general ecological principles as demonstrated by previous field work.

Equipment: maps showing the location of the oasis. equipment for studying plant and animal life in a location (See Chapter Four.)

Strategy

Question: What are the main features of an oasis ecosystem?

1. Trace outlines of the area of the oasis from an appropriate (large scale) map. If necessary scale-up the outlines to foolscap size.

2. Assign each group of students a specific activity e.g. studying plant distribution, comparing soil samples (and organisms) from locations at different distances from the water supply, studying the effects of human settlement, identifying food chains involving wild and domesticated organisms (including man) and comparing climatic features of the oasis with arid areas, over different times during a year and between night and day.

3. Students to produce an exhibit illustrating their study and present it to the rest of a class.

4. Discuss the connections between the studies.

5. Attempt to answer the questions: To what extent do people living in an oasis appear to understand its ecology? How can an oasis ecosystem be maintained?

8. STUDIES OF THE ECOLOGY OF INDIVIDUAL ORGANISMS (ANTECOLOGY)

(A, B; I. II)

Plants

Ecology can be approached through a number of perspectives. In particular one can distinguish between studies which consider whole communities and their environments (synecology) from those in which the relationships of a single species (autecology) are investigated. It is valuable to include some autecological studies in an ecology course.

In arid areas a useful collection of plants to be studied from this perspective would be a tree e.g. date palm, a succulent e.g. Zygophyllum simplex, a true xerophyte e.g. Calamagrostus arenaria and a water plant from a dam or irrigation canal e.g. water hyancinth.

Objectives

1. To understand the ecological relations of individual species.
2. to appreciate the relation between an organisms adaptations and its ecological associations.

Curriculum links

Adaptation, distribution, conservation, parasitism, reproduction.

Date Palm Tree

Preparation

Teacher: If necessary obtain permission to work on palm trees. (They are often owned by someone.) Arrange students in groups for various activities.

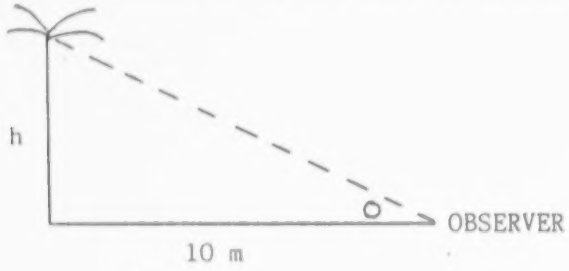
Students: Revise pollination and fertilisation. Revise use of sin formula.

Equipment: soil borer
apparatus for determing soil moisture content (See Chapter Four)

Strategy

1. Draw a map of the area showing distribution of trees and other plants.

2. Approximate the height of some trees through geometrical methods i.e. estimate the angle from the ground to the top of the tree whilst standing, say, 10 metres away.



The height of the tree (h) is then determined by the formula

h = length of tree from observer x sin of angle of observer to top of tree

From diagram h = 10 x sin O

3. Obtain soil and small root samples at different depths around the trees.

4. Relate the soil moisture content to the height of the trees and the extent of their root systems.

5. If possible obtain samples of flowers and fruits. (It can be dangerous for students to climb trees.)

6. Determine the mode of pollination and fertilisation of the plants. How does this affect distribution? (Remember some date palms are planted.)

7. Look for small trees sprouting out of large ones. Enquire how these are transplanted.

8. Find out if there are any insects on the palm leaves. If you see black spots, look for insects by mounting one of the spots, on a slide. Use a microscope to identify the insects. (The insects are called aphids. Examine several leaves and determine the degree of infection.)

9. After removing the aphid, examine its place on the leaf. Determine how aphids feed.

10. Observe any larger animals feeding on (or associated with) the palm trees.

11. Construct diagrams and discuss the ecosystem based on the palm trees.

Follow up

Study the management of palm tree plantations.

A Succulent (Zygophyllum simplex)

Preparation

Teacher: Arrange students in groups of 2 or 3. Half the groups in a class could do this activity whilst the other half were doing the next activity using Calamagrostus arenaria or a similar plant.

Students: Revise stem and root morphology.

Equipment: small hoe
hand lense and/or microscope
sharp knife or razor

Strategy

1. Map the distribution of the plant in the area being studied.

2. Dig up a few plants and compare the length of their roots with that of their stems. Explain the ecological reasons for the differences in length.

3. Feel the stem and the leaves. Explain the reasons for their particular texture.

4. Make a labeled diagram of the different parts of the plant.

5. Cut transvere sections of the stem and roots and examine under a lense or miscroscope. Explain the functions of parts observed.

6. Note any animals found on or near to the plants.

7. Construct diagrams and discuss the ecosystem based on the succulents.

Follow up (or in parallel)

1. Repeat this activity using a true xerophyte such as Calamagrostus arenaria or a similar plant. Attempt to explain the similarities and differences. Figures 48 and 49 , page 263 and 264.

2. Undertake studies of osmotic relations of the cells of both these plants and explain the ecological significance.

FIGURE 48

ZYGOPHYLLUM SIMPLEX

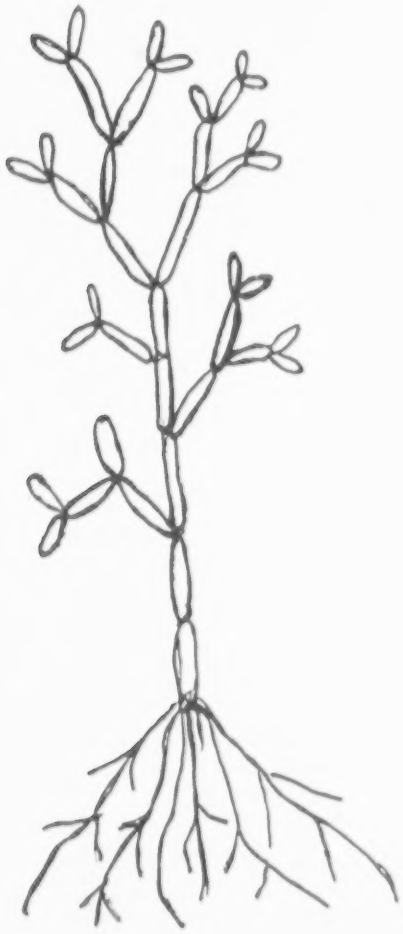


FIGURE 49

CALAMAGROSTUS ARENARIA

Water Hyacinth

Preparation

Teacher: Arrange for students to observe water hyacinths in a canal or other location and collect specimens for examination in school. As the water hyacinth is a pest the number collected need not be limited.

Students: Revise plant structure and transpiration.

Equipment: Hand lens and/or microscope
slides
sharp knife or razor
two large beakers or bowls

Strategy

1. Note the areas where plants are more and less distributed. Propose explanations.

2. Examine the external structure of the leaves and roots of the plant. Feel the texture. How is the plant adapted to its environment?

3. Cut sections of the leaf and examine using magnification. What adaptive features are observed?

4. Place plants in a bowl and mark the level of the water. Fill another bowl of the same size with water to the same level. Compare the rates of loss of water from the two bowls in cms. decrease in height per two hourly period. Explain the results.

Follow up

1. Organise long-term studies of the rate of area of growth of the plant. Note the influence of lack of herbivores and other inhibitors.

2. Study the adverse effects of the spread of the water hyacinth both locally and in other areas. Discuss methods employed to control it.

9. STUDIES OF THE BEHAVIOUR AND ECOLOGY OF DESERT ANIMALS

Chapter Five and several activities in other sections of this chapter provide details of techniques for studying animal behaviour and its relation to ecology. They should be consulted in order to devise activities relevant to local circumstances based on the ideas outlined here.

The Camel

The camel is often known as the ship of the desert. It is well-known for its ability to go for long periods without water. There are two main camel species: the one-humped common in African and Arabian deserts, and the two-humped which is common in Asian deserts. Incidentally, the camel can withstand cold as readily as heat. The two-humped camel survives the blizzards in Asia.

Camels may be found in your neighbourhood. Otherwise it might be possible to study them in a zoo.

Topics for study

1. Examine features of the camel which demonstrate its adaptation to desert conditions e.g. the upper lip, eyelashes, nostrils, neck, hump and feet.

2. Observe feeding habits and movement; link these to their ecology.

3. Develop the idea of camels as distributors of other organisms through their feeding habits linked to subsequent defaecation.

4. Obtain information about the habits and management of camels from their owners.

Goats

Goats are common in semi-arid zones being kept by man as a major source of meat, milk and wool. The domesticated varieties are different from their wild cousins in several characteristics such as: size, sorus, weight, and their ability to climb.

Topics for Study

1. Grazing habits. For example, locate a grazing area where plants grow wild around a source of water. Make arrangements with a shephard to have his goats feed in this area during the time students are at the location. Have students observe particularly how goats deal with the root system of plants. Compare grazed area with one where there is no grazing. Link to experiments on the effect of grazing based on Activities 13 and 14 of the Savanna section of this chapter.

2. Other destructive behaviour of goats and its relation to the role of these animals in bringing about desertification.

3. Devise strategies for regulating the management of goat-keeping.

Lizards

Lizards, of a variety of species, are relatively common in arid or semi-arid habitats. They can also be kept in terraria in which the environment they are to be found in is copied as closely as possible. They are not difficult to catch being attracted by meat or insect bait set in a container which is then closed by the observer directly or by a spring-loaded device. Lizards can also sometimes be obtained from dealers.

Topics for Study

1. Territoriality. With food being scarce in the desert it is important that animals are spaced out. Lizards have specific behaviour patterns to facilitate this.

2. Water retention. Comparisons of water content of food and faeces can be undertaken. Urination is infrequent.

3. Adaptation to arid conditions. Burrowing, pastures, behaviour patterns night and day.

4. Feeding habits. Food capture. Position in food web.

5. Breeding habits. Numbers of eggs, care of eggs, survival of young.

Locusts

Locusts are a wide-spread pest responsible for destroying crops in many parts of the world. They will be found in vegetation and can be collected using a sweep net. (See Chapter Four, Figure 6, page 49)

Topics for Study

1. Feeding habits. Examine mouth parts and their function.

2. Food consumption. Determine amount of vegetation consumed per individual (or a small number) and extrapolate to assess the effects of large populations. This provides an introduction to the study of food chains in relation to population size and food cycles. Locusts are preyed on by birds, lizards and other animals.

3. Life cycle. Locusts can be bred in captivity and estimates of net reproduction rate may be obtained.

4. Migration. Occasionally observations of the mass migration of locusts may be possible. This can provide the opportunity to consider factors affecting their movement and the ecological effects of it.

Water Snails

Diseases such as Bilharzia are carried by water snails and, thus, knowledge of their behaviour and ecology is crucial in order to control the spread of the diseases. Snails are to be found in canals, irrigation ditches and similar locations. The water is usually clear and preliminary observation of, for example, the distribution of the snails in relation to water plants can be made. However, more detailed studies are best done in an aquarium.

Care needs to be taken in handling water in which snails have been present. Bilharzia is contracted from free-swimming larvae (cercariae) in the water, not directly from the snails. To be safe use rubber gloves.

Topics for Study

1. Distribution of snail, movement and feeding habits.

2. Food chains of the snail and its parasites.

3. Methods of control of snail population and disease transmission.

BIBLIOGRAPHY

Ayensu, E. S. and Manton-Lefevre, J. (Edit.) (1981) Proceedings of the Symposium on the State of Biology in Africa. ICSU/UNESCO.

Baez, A. V., Knamiller, G. W. and Smyth, J. C. (Edit.) (1987) The Environment and Science and Technology ducation. Pergamon Press, Oxford. ICSU Press, Paris.

Batanouny, K. H. (1981) Ecology and Flora of Qatar. Center of Scientific and Applied Research, University of Qatar.

Bennett, D. P. and Humphries, D. A. (1977) Introdution to Field Biology. 2nd edition. Edward Arnold, London.

Bouliere, F. (Ed.) (1983) Tropical Savannas. Ecosystems of the World 13. Elsevier, Amsterdam.

Bouliere, F. (1987) Tropical Ecology: its Beginnings, Specificity and Constraints. Biology International, No. 15. IUBS, Paris.

Brown, A. L. (1978) Ecology of Soil Organisms. Heinemann Education Books. London.

Burton, J. (1979) Animals of the African Year - The Ecology of East Africa. Peter Lowe, London.

Crutch, J. W. (1980) Voice of the Desert: The Naturalist Interpretation. Apella Books, U.S.A.

Dicastri, F., Baker, F. W. G. and Hadley, M. (1984) Ecology in Practice. Part 1 Ecosystem Management. Part 2 The Social Response. Tycooly International,Dublin. UNESCO, Paris.

Dowdeswell, W. H. (1967) Practical Animal Ecology. Methuen, London.

Huntley, B. J. and Walker, (Eds.) (1982) Ecology of Tropical Savannas. Springer-Verlag, New York.

Jenkins, P. F. (1976) School Grounds - Some Ecological Enquiries. Heinemann Educational Books, London.

Kelly, P. J. and Schaefer, G. (Edit.) (1980) Biology Education for Community Development. Taylor and Francis, London.

Kershaw, K. A. (1973) Quantitative and Dynamic Plant Ecology. Edward Arnold, London.

Kormondy, E. (1982) Conception cology. Prentice Hall Inc.

Lamotte, M. and Hadley, M. (1984) Biosphere reserves in savanna regions: building the research conservation connection. Conservation, Science and Society. UNESCO-UNEP National Resources Research XXI, UNESCO, Paris.

Maury-Lechon, G., Hadley, M. and Younes, T. (Edit.) (1984) The Significance of Species Diversity in Tropical Forest Ecosystems. Biology International Special Issue - 6. IUBS, Paris.

Murray, S. and Louis, B. (1980) Investigations in Modern Biology. Cambridge Books Inc., New York.

Pomeroy, D. E., Servie, M. W. and Finner, M. (1984) Applied Ecology for Tropical Africa. Longman.

Raven, P. H. and Johnson, G. (1986) Biology. Mosly College Publishing Inc., U.S.A.

Sands, M. K. (1978) Problems in Ecology. Mills and Boon, London.

Solbrig, O. T. and Golley, F. B. (1983) A Decade of the Tropics. Biology International Special Issue - 2. IUBS, Paris.

Spellerberg, I. F. and Pritchard, A. J. (1984) Ecology, Ecosystem Management and Biology Teaching. UNESCO, Paris.

Swift, M. J. (1987) Tropical Soil Biology and Fertility. Biology International Special Issue - 13. IUBS, Paris.

Symoens, J. J. et al. (Eds.) (1981) The Ecology and Utilization of African Inland Waters. UNEP, Nairobi.

UNESCO (1979) Tropical Grazing Land Ecosystems. A state-of-knowledge report prepared by UNESCO/UNEP/FAO. Natural Resources Research XXVI, UNESCO, Paris.

Wallwork, J. A. (1970) Ecology of Soil Animals. McGraw Hill, London.

INTERNATIONAL PROGRAMMES CONCERNED WITH ECOLOGY TEACHING

Decade of the Tropics Programme	International Union of Biological Sciences, 51 Boulevard de Montmorency, 575016 Paris, FRANCE.
Division of Science, Technical and Environmental Education	UNESCO, 75700 Paris, FRANCE.
Man and the Biosphere Programme (MAB)	UNESCO, 75700 Paris, FRANCE.
United Nations Environmental Programme (UNEP)	Environmental Education and Training Unit, Nairobi, Kenya.
World Wildlife Fund (WWF) and International Union for Conservation of Nature and National Resources (IUCN)	Avenue du Mont-Blanc, 1196, Gland, SWITZERLAND.
WWF/IUCN International Eduction Project	Greenfield House, Guiting Power, Gloucestershire, GL54 5TZ, U.K.